AD-A175 168 STUDIES IN PHLEBOTOMINE SAND FLIES(U) FLORIDA UNIV GAINESVILLE DEPT OF ENTOMOLOGY AND NEMATOLOGY D G YOUNG 30 JUN 82 DADA17-72-C-2139 1/1

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STUDIES OF PHLEBOTOMINE SAND FLIES

Annual Report

by

D.G. Young

Department of Entomology and Nematology
University of Florida

June 30, 1982

Supported by

U.S. Army Medical Research and Development Command
Fort Detrick, Frederick, Maryland 21701

Contract No. DADA17-72-C-2139

University of Florida
Gainesville, Florida 32611

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SECURITY CLASSIFICATION OF THIS PAGE

ADA175168

REPORT DOCUMENTATION PAGE

Form Approved
OMB No 0704-0188
Exp. Date Jun 30, 1986

1a. REPORT SECURITY CLASSIFICATION Unclassified	1b. RESTRICTIVE MARKINGS
2a. SECURITY CLASSIFICATION AUTHORITY	3. DISTRIBUTION/AVAILABILITY OF REPORT Approved for public release; distribution unlimited
2b. DECLASSIFICATION/DOWNGRADING SCHEDULE	
4. PERFORMING ORGANIZATION REPORT NUMBER(S)	5. MONITORING ORGANIZATION REPORT NUMBER(S)
6a. NAME OF PERFORMING ORGANIZATION University of Florida	6b. OFFICE SYMBOL (If applicable)
7a. NAME OF MONITORING ORGANIZATION	
6c. ADDRESS (City, State, and ZIP Code) Gainesville, Florida 32611	7b. ADDRESS (City, State, and ZIP Code)
8a. NAME OF FUNDING/SPONSORING ORGANIZATION U.S. Army Medical Research & Development Command	8b. OFFICE SYMBOL (If applicable)
9. PROCUREMENT INSTRUMENT IDENTIFICATION NUMBER DADA17-72-C-2139	
8c. ADDRESS (City, State, and ZIP Code) Fort Detrick, Frederick, MD 21701-5012	10. SOURCE OF FUNDING NUMBERS

PROGRAM ELEMENT NO.	PROJECT NO.	TASK NO.	WORK UNIT ACCESSION NO.
62770A	3M162.770A870	AC	009

11. TITLE (Include Security Classification)
(U) Studies of Phlebotomine Sand Flies

12. PERSONAL AUTHOR(S)
D. G. Young

13a. TYPE OF REPORT	13b. TIME COVERED	14. DATE OF REPORT (Year, Month, Day)	15. PAGE COUNT
Annual Report	FROM 6/30/81 TO 6/30/82	30 June 1982	

16. SUPPLEMENTARY NOTATION

17. COSATI CODES		
FIELD	GROUP	SUB-GROUP

18. SUBJECT TERMS (Continue on reverse if necessary and identify by block number)
Lutzomyia Leishmaniasis
Phlebotomus
Sand Fly

19. ABSTRACT (Continue on reverse if necessary and identify by block number)

Phlebotomus martini, a vector of visceral leishmaniasis in Kenya, was colonized in the laboratory for the first time using larval food and other techniques developed at the University of Florida. Phlebotomus duboscqi, a vector of cutaneous leishmaniasis, was developed in Kenya at a site where Leishmania major has been isolated from rodents. A paper describing the previously unknown male of Sergentomyia blossi from Kenya was completed. The eggs of 5 Lutzomyia species were compared with the aid of the scanning electron microscope. Specific differences in the outer structure of the eggs were found. A new species of Lutzomyia from California and 2 new species from Arizona were discovered. They and 11 other North American Lutzomyia were illustrated in a review paper of the North American fauna. Additional species, including 2 undescribed taxa, were studied from Ecuador, Peru and Surinam. The reference collection of New World Phlebotominae now contains specimens of 280 American species with 66 holotypes and nearly 100 paratypes making it one of the most complete collections in existence. Five Lutzomyia species from the U.S.A. are being maintained in the laboratory; 3 of them, L. shannoni, L. diabolica and L. anthrophora transmitted (Continued)

20. DISTRIBUTION/AVAILABILITY OF ABSTRACT ☐ UNCLASSIFIED/UNLIMITED ☒ SAME AS RPT. ☐ DTIC USERS	21. ABSTRACT SECURITY CLASSIFICATION
22a. NAME OF RESPONSIBLE INDIVIDUAL Mrs. Judy Pawlus	22b. TELEPHONE (Include Area Code) 301-663-7325 / 22c. OFFICE SYMBOL SGRD-RMI-S

DD FORM 1473, 84 MAR 83 APR edition may be used until exhausted. All other editions are obsolete.

SECURITY CLASSIFICATION OF THIS PAGE

#19 Abstract (Continued)

Leishmania mexicana (Texas strain) to hamsters in the laboratory as early as 4 days following the infecting bloodmeal.

SUMMARY

Phlebotomus martini, a vector of visceral leishmaniasis in Kenya, was colonized in the laboratory for the first time using larval food and other techniques developed at the University of Florida. *Phlebotomus duboscqi*, a vector of cutaneous leishmaniasis, was discovered in Kenya at a site where *Leishmania major* has been isolated from rodents. A paper describing the previously unknown male of *Sergentomyia blossi* from Kenya was completed. The eggs of 5 *Lutzomyia* species were compared with the aid of the scanning electron microscope. Specific differences in the outer structure of the eggs were found. A new species of *Lutzomyia* from California and 2 new species from Arizona were discovered. They and 11 other North American *Lutzomyia* were illustrated in a review paper of the North American fauna. Additional species, including 2 undescribed taxa, were studied from Ecuador, Peru and Surinam. The reference collection of New World Phlebotominae now contains specimens of 280 American species with 66 holotypes and nearly 100 paratypes making it one of the most complete collections in existence. Five *Lutzomyia* species from the U.S.A. are being maintained in the laboratory; 3 of them, *L. shannoni*, *L. diabolica* and *L. anthophora* transmitted *Leishmania mexicana* (Texas strain) to hamsters in the laboratory as early as 4 days following the infecting bloodmeal.

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PROGRESS REPORT

A. INTRODUCTION

The importance of phlebotomine sand flies as vectors of leishmaniasis and arboviruses in many parts of the world is considerable. The World Health Organization estimates that 400,000 new human cases of leishmaniasis occur each year in the world. At present, there is an epidemic of visceral leishmaniasis (kala-azar) in India. Over 100,000 human cases have been reported. In Iran the incidence of cutaneous leishmaniasis has risen sharply due to the movement of soldiers and other nonimmunes in endemic foci. In the Isfahan and Khuzistan areas, the incidence of the disease is estimated to be 30 cases per 100 people, total population at risk is about 100,000 persons. In Peru there is currently a resurgence of cutaneous leishmaniasis and bartonellosis in Andean valleys; 495 cases of the latter disease were reported in the entire country in 1981. It is believed that over 3,000 cases will be reported in 1982 based on present data.

The WHO in 1977 stated that "...taxonomic studies of the vectors are of fundamental importance due to the difficulty in identifying them." The WHO Scientific Working Group on the Leishmaniases recommended that continued support be given to maintaining sand fly reference collections and to projects dealing with the systematics of the group. There is a need for new practical field keys. The establishment of laboratory colonies of vectors is considered important for studies on genetics and vector competence.

B. OBJECTIVES

The objectives of this project are:

1) To prepare keys, illustrations and other aids to identification both by geographic areas and by taxonomic groups.

2) To arrive at a more satisfactory classification of the subfamily Phlebotominae.

3) To build a reference collection of Phlebotominae.

4) To maintain one or more species in the laboratory to provide immatures and adults for taxonomic and disease studies.

C. RESULTS

Kenya trip (16 June - 7 Aug. 1981). Specific results on studies of phlebotomine sand flies in Kenya are given in Appendix I. The most significant accomplishment was the establishment and successful colonization of *Phlebotomus martini* in the laboratory. This species is the suspected vector of visceral leishmaniasis in that country and has not been previously colonized in the laboratory. A paper reporting this accomplishment is in press (Beach et al. 1983).

The PI and Captain Ray F. Beach discovered *Phlebotomus duboscqi* at Marigat, Baringo District. This species, belonging in the medically important subgenus *Phlebotomus* and previously unknown in Kenya, is a proven vector of cutaneous leishmaniasis elsewhere in Africa. *Leishmania* isolated from rodents from Baringo were earlier identified as *Leish. major*--the parasite that is transmitted by *P. duboscqi* in west and central Africa. Thus, the possibility exists that this sand fly may be involved with transmission of this parasite among rodents in the Baringo District as well. A short paper reporting the discovery of *P. duboscqi* in Kenya was submitted for publication (Beach et al. 1982).

The male of *Sergentomyia blossi*, also previously unknown (undescribed), was discovered by the PI during this trip. A paper describing it was accepted for publication (Young et al. 1982).

Taxonomy of New World Phlebotomines. Eggs of five *Lutzomyia* spp. were studied with the aid of the scanning electron microscope (Appendix II). The existing classification of eggs, based on the egg surface structure, was slightly modified although too few species are represented and thus the classification

remains tentative.

A paper describing a new species of Lutzomyia from California was completed and sent to press (Young et al. 1983). Unlike most other New World phlebotomines, this species occurs in arid habitats.

Two additional undescribed species of Lutzomyia from the U.S.A. were discovered. Descriptions and illustrations of them and all other North American species were completed. A review of this fauna, including identification keys, is given in Appendix III.

Flight trap collections in Ecuador in 1981 and 1982 yielded another new species of Lutzomyia (Trichophoromyia) as well as L. ayrozai, an anthropophilic species previously unreported in Ecuador. The specimens were given to the PI by colleagues working in the country. Other newly-collected specimens from Peru, Brazil and Surinam were examined. Lutzomyia (vexator group) n. sp. and L. shawi were taken in man-biting collections in Amazonas, Peru. Both represent new records for the country. These and other species were illustrated and will be included in the handbook of the neotropical phlebotomines which is presently being compiled.

The reference collection continued to grow with over 280 American species represented, making the collection one of the most complete in existence. Sixty-six holotypes and nearly 100 paratypes are included in this collection.

Laboratory colonization of phlebotomines. Five species of Lutzomyia, L. vexator, L. diabolica, L. anthophora, L. shannoni and L. cruciata, are being maintained in our laboratory. The paper on rearing techniques, mentioned in the 1981 Annual Report, is now in press (Endris et al. 1982). So far, attempts to rear 9 out of 9 phlebotomine species using these methods, developed in our laboratory, have been successful.

A trip to Texas in May and June 1982 was made to obtain living material of L. diabolica. Six hundred females were dissected after oviposition to search

for natural infections of *Leishmania*. None was positive but enough eggs were obtained to initiate a strong laboratory colony. This species is strongly anthropophilic and is the suspected vector of cutaneous leishmaniasis in Texas and Northern Mexico (Appendix III). This species, *L. shannoni* and *L. anthophora*, all of which occur in the U.S.A., are capable of transmitting *Leishmania mexicana* (Texas strain) to hamsters in the laboratory from 4 to 6 days following an infecting bloodmeal. The vector potential of their species was previously unknown.

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PERSONNEL SUPPORTED ON PROJECT

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S.S. Haney, Technician, Dept. of Entomology and Nematology, University of Florida.

APPENDIX I

Results of Studies on Phlebotomine Sand Flies in

Kenya (16 June - 7 August 1981)[1]

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Introduction

The role of sand flies as vectors of cutaneous and visceral leishmaniasis in Kenya and elsewhere is well known. *Phlebotomus* (*Synphlebotomus*) *martini* Parrot is the incriminated vector of Kala-azar in Kenya but the female is morphologically inseparable from those of *P. celiae* Minter and *P. vansomerenae* Heisch, Guggisberg & Teesdale. Thus, the specific identification of wild-caught females in some endemic areas of Kenya is open to question.

In order to study possible differences among these taxa and to furnish material for experimental work, it is considered important to establish laboratory colonies of these species. In addition to this objective, fresh material of other species is required to complete an identification key to the sand flies of Kenya. The present distribution of phlebotomines requires further study.

The following work, considered to be a preliminary effort, was conducted by

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[1]Scientist - in Residence, Leishmaniasis Vector Biology, The International Centre of Insect Physiology and Ecology, Nairobi, 16 June - 1 August 1981. Financial support was provided by U.S. Army Medical Research & Development Contract DADA-17-72- C -2139.

three investigators - Dr. Mutuku Mutinga, ICIPE; Dr. Raymond F. Beach, U.S.A. Medical Research Unit - Kenya; and the author.

Methods and Materials

1. Field Collections.

Localities Sampled

A. Kalawa, Machakos District; various dates from 18 June - 24 July, 1981.

B. Tseikuru, Kitui District; 23 - 26 June.

C. Marigat, Baringo District (Perkerra Horticultural Sub-station); 7 - 10 July.

D. Rabai, Mombassa District; 24 - 28 July.

E. Amboselli National Park, Kajiado District; 7 August.

We aspirated sand flies from termite hills or other resting sites at each locality using a simple suction tube. At Marigat, human and calf bait were used to secure living flies. The insects were gently blown into a 230 ml plastic jar, the bottom fourth of which was filled with plaster of Paris and wetted. A plastic screen covered a 35 mm diameter hole in the lid, allowing for ventilation and for providing a surface for depositing drops of honey water. At the field laboratory, all flies collected during the day were released into a rectangular plexiglass feeding cage. A cloth sleeve covered one end. The bottom and back of this cage were covered with plaster of Paris to serve as a resting surface for the insects. To maintain high humidity, we placed the feeding cage inside a larger cooler chest with damp towels. Virtually all of these activities apply to the Marigat site where sand flies were relatively abundant.

A confined hamster and/or lizard were placed in the cage for at least two hours each day. Engorged female flies were then placed in individual 7 dr vials

and transported to Nairobi.

We also collected flies with CDC light traps and a flight trap. Dead flies were preserved dry between sheets of tissue paper in pill boxes.

2. Laboratory Techniques

We reared larvae following the procedures of Young et al. (1981). Laboratory methods for handling and feeding adults were essentially the same as those outlined above under "Field Collections."

Slide-mounting procedures followed those of Young (1979). Through the kindness of Dr. Mutinga, I had the opportunity to study some of the phlebotomines in his extensive collection at ICIPE.

Results

At Marigat, over 50% of unfed females in the genera *Sergentomyia* and *Phlebotomus* fed on the confined hamster or lizard. Mortality was surprisingly low and we secured over 300 eggs from 9 females of *P. martini* and hundreds of eggs from 3 spp. of *Sergentomyia* - *S. africana*, *S. schwetzi* and *S. antennata*. The fact that wild *martini* females fed readily on a confined hamster was especially encouraging. From these 300 eggs it appears that, for the first time, a laboratory colony of *P. martini* has been established (R. Beach, Personal comm.).

Taxonomic studies yielded several interesting results. Near Rabai, we collected a male of *Sergentomyia blossi*, previously unknown. A paper describing it has been submitted for publication (Young et al. 1982).

At Marigat in the Rift Valley where Kala-azar is endemic, we collected a male of *Phlebotomus* (*Phlebotomus*) *duboscqui* Neveu-Lemaire in a light trap (7-10 July). This discovery represents the first time this species, or any other in

the medically important subgenus Phlebotomus, has been found in Kenya. Elsewhere in Africa P. duboscqui is a vector of cutaneous leishmaniasis. Females are anthropophilic and will bite in the daytime inside houses.

In the "to be identified" section of Dr. Mutinga's collection, we identified two females of P. orientalis or close ally also from Marigat. Previous to this discovery, this species in Kenya was known only from two specimens collected in East Kenya outside of the Rift Valley. Males of this species are needed to confirm the identification. Its role in disease transmission, if any, remains to be determined. In the Sudan P. orientalis is a major vector of Kala-azar.

More than 1000 additional phlebotomines were collected, mostly common forms such as S. bedfordi, S. antennata, S. africana, S. schwetzi, S. suberecta and P. martini. Drawings of some of them were completed for use in an identification key being written. This work is continuing.

Although there was little time for comparison with light traps, the flight trap (Gressitt & Gressitt 1962) captured a fair number of sand flies at Marigat (142 specimens, 3 trap days). P. martini, S. bedfordi, S. antennata, S. schwetzi, S adleri & S. africana were represented. Traps of this kind can be left in the field for an indefinite period of time and are useful for studying seasonal distribution, direction of flight, etc.

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APPENDIX II (In press)

A COMPARISON OF OOCYTE TOPOGRAPHY OF FIVE PHLEBOTOMINE SANDFLIES (LUTZOMYIA) WITH THE SCANNING ELECTRON MICROSCOPE (DIPTERA: PSYCHODIDAE)[1]

INTRODUCTION

The egg surface structure of 19 neotropical Phlebotomine species has been described (Zimmerman et al. 1977, Ward Ready 1975) using the scanning electron microscope (SEM). Ward and Ready (1975) noted three species-specific topographic patterns, i.e., polygonal, parallel ridging, and volcano-like. Several authors (Chaniotis and Anderson 1964, Addis 1945, Lindquist 1936, Barreto 1941, and Sherlock 1957a,b, 1963) described and figured the eggs of 17 neotropical sandfly species using light microscopy. After examining the literature cited and eggs from the 5 species described herein we propose adding another category to the patterns of Ward and Ready (1975); that is, parallel ridges connected or parallel ridges unconnected.

The classification of eggs of 41 species of New World sandflies according to the proposed scheme is presented in Table 1.

MATERIALS AND METHODS

Eggs were obtained from females reared in laboratory colonies. The preparation method of eggs for SEM based on the work of Quattlebaum and Carner (1980) is as follows:

1. Eggs were placed on a filter paper disc in a 1 cm deep plastic container cut from a plastic film cannister.

2. The plastic container was floated in a 50 ml Tri-pour® polystyrene beaker containing 5 ml aqueous 1% OsO_4

3. The paper lid was installed and the entire container was sealed in Parafilm® and held in an exhaust hood at room temperature for 5 days.

4. After 5 days exposure to osmium vapor the inner container was transferred to a covered petri dish for 24 hr to allow slow drying of the eggs.

5. Eggs were attached to an SEM stub using either double-sided tape or 0.1% aqueous hydrobromide polylysine (Polysciences, Inc., Warrington, PA 18976), sputter-coated with approximately 300A°of gold in an Eiko Engineering IB-2 Ion Coater, and examined in a Hitachi S-450 scanning electron microscope (SEM) at 20 KV.

Eggs were measured in microns at 100x with a compound microscope and an ocular micrometer. Intact fresh eggs or recently hatched eggs were acceptable whereas old eggs or infertile eggs usually collapsed making accurate measurement difficult. Eggs to be measured were placed on a microscope slide in Histocon®. In each sample the eggs were produced by 5-10 females.

RESULTS

The SEM micrographs of the sandfly eggs are shown in Figures 1 and 2. Descriptions of eggs of each species are as follows: Measurements given are the range, mean, and standard deviation for egg length and width for each species.

Lutzomyia shannoni (Dyar, 1929), Florida specimens

Fig.1(2), 2(2)

Size: N = 102, L: 290-340 (330 ± 10), W: 70-110 (90 ± 10)

Exochorion: High, narrow longitudinal ridges connected by prominent perpendicular ridges forming 4 and 5 sided polygons which are frequently rectangular.

Lutzomyia diabolica (Young and Perkins 1982), Uvalde Co., Texas

Fig.1(1), 2(2)

Size: N = 47, L: 340-370 (350 ± 10), W: 90-110 (100 ± 10)

Exochorion: Surface topography is characterized by a series of discontinuous parallel longitudinal ridges that are not laterally connected.

Lutzomyia vexator (Coquillett 1907), Levy Co., Florida

Fig.1(3), 2(3)

Size: N = 193, L: 330-390 (380 ± 10), W: 80-110 (100 ± 10)

Exchorion: Surface topography consists of delicate parallel longitudinal ridges with regular perpendicular connections that form polygons which are nearly square. There are also occasional oblong cells.

Lutzomyia anthophora (Addis 1945), Cameron Co., Texas

Fig.2(5)

Size: N = 100, L: 330-370 (340 ± 10), W: 80-100 (80 ± 10)

Exochorion: Reticulation consists of weak parallel longtitudinal ridges with slight perpendicular connections at irregular intervals.

Lutzomyia cruciata spp. (Young and Perkins 1982), Alachua Co., Florida

Fig.1(4), 2(4)

Size: N = 61, L: 320-370 (340 ± 10), W: 80-120 (100 ± 10)

Exochorion: Wide, flat, parallel longitudinal ridges with occasional weaker connecting ridges which are not usually perpendicular to the longtitudinal ridges.

Discussion

Several techniques were tried for preserving the eggs to prevent collapse under vacuum in the SEM column. The method used here yielded the best results when fertile eggs were used.

A "standard" EM fixation procedure using 1% OsO_4 as a fixative followed by 5% aqueous acrolein, dehydration in dimethoxpropane and acetone, then critical point drying with Freon as a transition solvent proved unsuccessful because most specimens collapsed in SEM. Lyophilization and critical point drying of eggs without fixation were also unsuccessful.

A technique which was not used but one which may be promising is freeze drying.

The size variation of eggs laid by individual females was determined by measuring 10 eggs from each of 10 L. vexator females. The variation in egg length and width between females ranged from 10-50 microns and from 10-30 microns, respectively. As a result of broad intraspecific variation it is not possible to separate the eggs of different sandfly species by size. Therefore, the surface sculpturing is the only characteristic of the egg that can be used for species determination.

Table 1. Classification of 41 species of neotropical phlebotomine sandfly eggs based on oocyte topographic patterns.

	TOPOGRAPHIC PATTERN			
Describer	Polygon	Parallel Ridges (connected)	Parallel Ridges (unconnected)	Volcano-Like
Endris et al.	*L. texana*	*L. cruciata* spp.	*L. diabolica*	
	L. vexator	*L. anthophora*		
	L. shannoni			
Sherlock	*L. lenti*		*L. renei*	
	L. bahiensis			
Zimmerman et al.	*L. sanguinaria*			
	L. trapidoi			
	L. ylephilator			
	L. gomezi			
Chaniotis	*L. vexator*			
	L. occidentalis			
Ward and Ready	*L. antunesi*		*L. longipalpis*	*L. flaviscutellata*
	L. yuilli			*L. complexa*
	L. nsp. 260.43 (= *L. shawi*)			*L. lainsoni*
	L. nsp. 260.44 (= *L. richardwardi*)			*L. davisi*
	L. dendrophyla			*L. paraensis*
	L. gomesi			
Barreto	*L. guimaraisi*	*L. pestanai*	*L. lanei*	
	L. pessoai	*L. arthuri*	*L. whitmani*	
	L. fischeri	*L. intermedia*	*L. alphabetica*	
	L. limai			
	L. monticola			

ACKNOWLEDGEMENTS

We thank E. Ann Ellis, Thelma Carlysle, and William Carpenter for their technical advice and assistance on specimen preparation and SEM.

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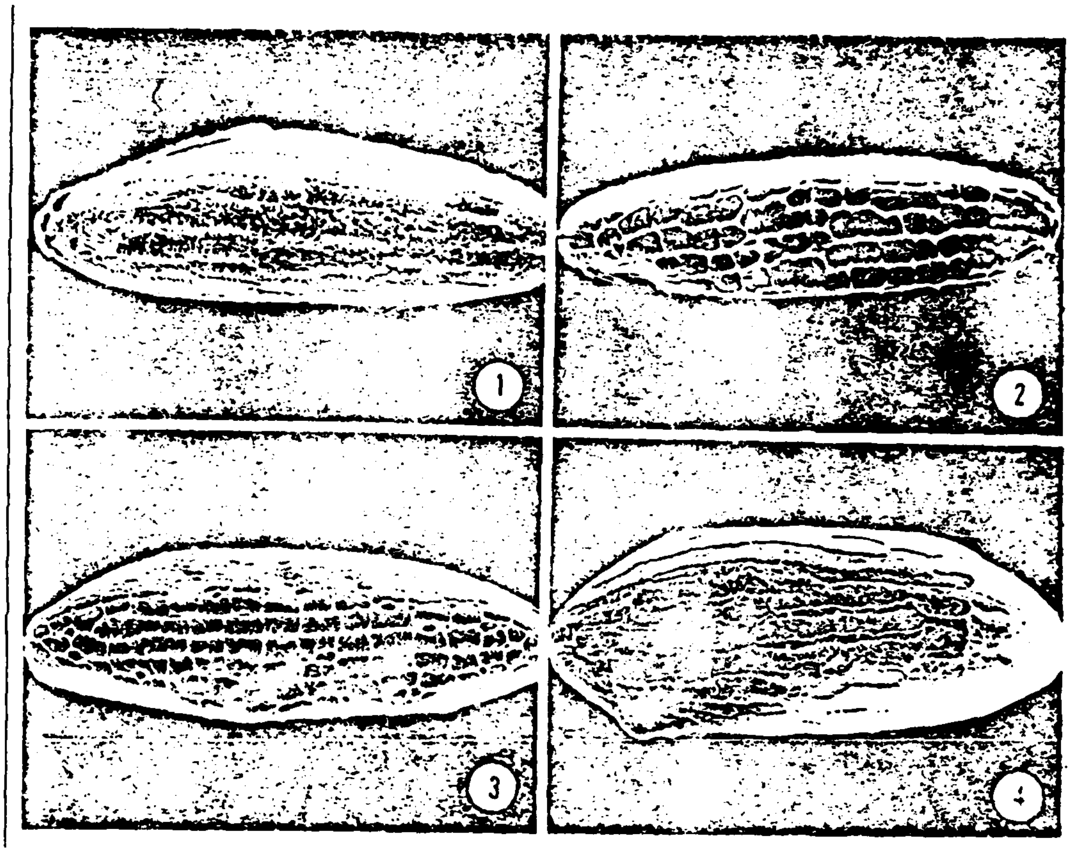


Figure 1. Scanning electron micrographs of eggs of four sandfly species. (1) Lutzomyia diabolica, (2) Lutzomyia shannoni, (3) Lutzomyia vexator, (4) Lutzomyia cruciata

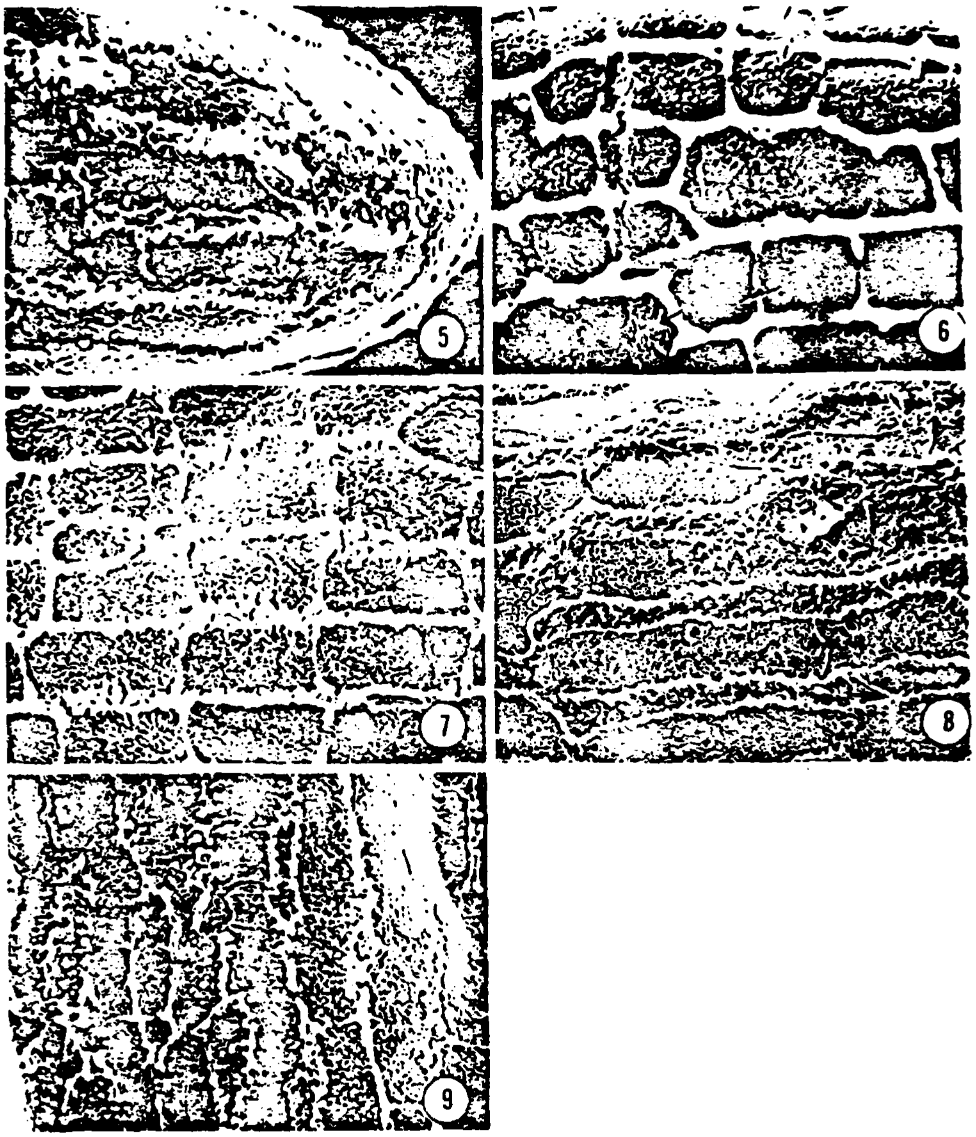


Figure 2. Scanning electron micrographs of oocyte topography of five species of sandfly. (5) Lutzomyia diabolica, (6) Lutzomyia shannoni, (7) Lutzomyia vexator, (8) Lutzomyia cruciata , (9) Lutzomyia anthophora.

Appendix III

ISSN 0027-142X

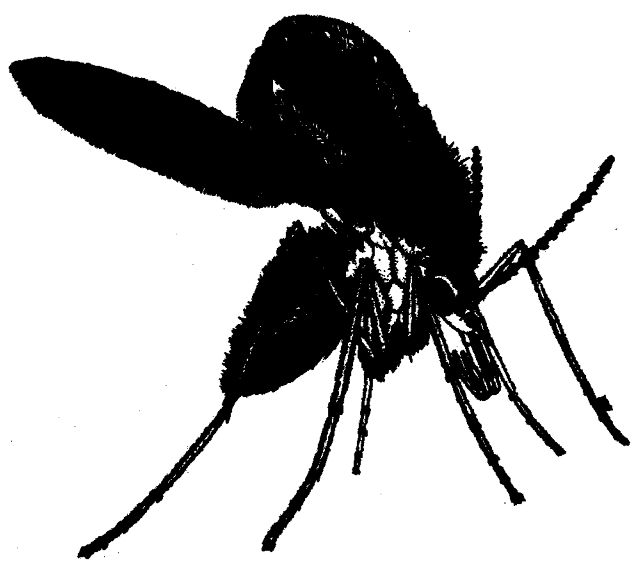
Mosquito News

JOURNAL OF THE
AMERICAN MOSQUITO CONTROL ASSOCIATION

VOLUME 44 | JUNE 1984 | NUMBER 2—PART 2

Phlebotomine Sand Flies of North America
(Diptera: Psychodidae)

D. G. Young and P. V. Perkins



A female phlebotomine sand fly

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JOURNAL OF THE AMERICAN MOSQUITO CONTROL ASSOCIATION

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Published on the 20th of March, June, September, and December by the

AMERICAN MOSQUITO CONTROL ASSOCIATION, INC.

T. D. MULHERN, *Executive Director*, 5545 East Shields Avenue. Fresno, California 93727. Tel. 209-292-5329

Printed by the Boyd Printing Co., Albany, NY 12210

SUBSCRIPTION RATES

$35.00 per year, Single copies $8.75

AMCA MEMBERSHIP DUES (Includes subscription)

$25.00 per year

For airmail overseas, add $15.00 per year or $3.75 per individual number.

PUBLICATION OFFICE: Room 3140, Cultural Education Ctr., Albany, NY 12230

EDITORIAL OFFICE: Dept. of Entomology, Walter Reed Army Institute of Research, Washington, DC 20307

BUSINESS OFFICE OF AMCA: 5545 East Shields Avenue, Fresno, California 93727

POSTMASTER: Send address changes to *Mosquito News*, 5545 East Shields Ave., Fresno, CA 93727

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Second class postage paid at Fresno, CA and additional mailing offices

PHLEBOTOMINE SAND FLIES OF NORTH AMERICA (DIPTERA: PSYCHODIDAE)[1]

D. G. YOUNG[2] AND P. V. PERKINS[2]

ABSTRACT. Fourteen species of *Lutzomyia* sand flies are recorded from North America, north of Mexico. Two of them, *L. apache* n.sp. and *L. tanyopsis* n.sp. from Arizona, are described for the first time. *Lutzomyia diabolica* is treated as a valid species, distinct from *L. cruciata*. *Lutzomyia vexator*, having a widespread geographic distribution in North America, is considered as a monotypic taxon without subspecies. Identification keys, distributional data, references, biological and disease information are provided.

INTRODUCTION

During the past decade, interest in North American sand flies has increased due to confirmed reports of autochthonous human leishmaniasis in Texas (Shaw et al. 1976), canine leishmaniasis in Oklahoma (Anderson et al. 1980), and Rio Grande virus in *Neotoma* woodrats in Texas (Calisher et al. 1977). Phlebotomines are the suspected, but as yet unproven, vectors of these diseases in these foci.

This report summarizes the published and unpublished information on the taxonomy and geographic distribution of the known species of *Lutzomyia* França in North America (north of Mexico), including information on their bionomics and vector potential where known. References and identification keys are provided.

Coquillett (1907) first reported sand flies in the Western Hemisphere. He described *Lutzomyia cruciata* (as *Flebotomus cruciatus*) from Guatemala and *L. vexator* (as *Flebotomus vexator*) from Plummer's Island, Maryland. Shannon (1913) observed *L. vexator* females feeding on snakes at the type-locality and speculated that another female had bitten a person sleeping inside of a cabin.

A group of biologists from Cornell University were tormented at night by biting sand flies on Minnie's Island, Okefenokee Swamp, Georgia in 1912. Johannsen (1943) recalled that a "member of the party counted over 75 punctures on his body, another's hands and arms were swollen to abnormal size. . . ." Local residents, acutely aware of these insects, called them "merye wings," an Elizabethan English term reflecting the origin of most of the residents. Johannsen (1943) studied one defective sand fly (lot no. 1981), concluding that it differed from *L. vexator* in wing venation and palpal proportions but he did not further describe the specimen which has since been lost (Quentin Wheeler, in litt., 1982). The identity of these man-biting sand flies remains unknown, but they probably represented *L. shannoni* (Dyar) and/or *L. cruciata*.

Apart from this report and from man-biting observations of *L. shannoni* in the southeastern USA, the only other area in North America where sand flies commonly bite people is in southcentral Texas. Parman (1919) observed a species, later named *Phlebotomus diabolicus* Hall 1936, biting people at Uvalde, Texas. Lindquist (1936) added more information on the habits of this species and reported that females attacked man at night in dwellings from 2000 to 2400 hr. We now know that *L. diabolica* will attack man in the daytime as well.

PHLEBOTOMINE-BORNE DISEASES IN THE UNITED STATES

Cutaneous leishmaniasis in humans

Simpson et al. (1968) documented the first parasitologically-confirmed human case of autochthonous leishmaniasis in the USA. Leishmaniae were observed in tissue smears and culture medium. The patient, who suffered from diffuse cutaneous leishmaniasis, had lived in San Benito, Cameron Co., Texas for her entire life except for occasional visits to the Mexican border states of Tamaulipas and Nuevo León. Subsequently, Shaw et al. (1976) discussed 2 additional human cases of cutaneous leishmaniasis in Texas (Gonzales and Dilworth counties). They believed that the disease had been locally acquired in both cases. Walton et al. (1977) identified one of the Texas strains (no. 1156) as belonging in the *Leishmania mexi-*

[1] Research supported by U.S. Army Medical Research and Development Command contracts DADA 17-72-C-2139 and DAMD 17-82-C-2223, and by the Steffen Brown Foundation. Fla. Agric. Exp. Stn. Journal Series No. 5347.

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cana complex. Peters (1981), referring to the Texas *Leishmania* as *Leish. mexicana pifanoi*, suggested that the disease may have been introduced from Venezuela. Kreutzer et al. (1983) identified one Texas strain (WR 127) as *Leishmania mexicana mexicana*. Parasites were cultured from a boy with a facial lesion, who probably contracted the disease in the vicinity of Uvalde, Texas in 1981 (Larry Hendricks, personal communication). Three unrelated persons from or near San Antonio, Texas, contracted cutaneous leishmaniasis in 1982 and it is believed the infections were also locally acquired (Tracy Gustafson, personal communication). In view of these reports, there is a good possibility that Stewart and Pilchner (1945) earlier had correctly diagnosed cutaneous leishmaniasis in a boy who lived near Alice, Texas.

The epidemiology of leishmaniasis in Texas, though not yet studied, may be similar to that in nearby Coahuila State, Mexico where Ramos-Aguirre (1970) discussed 3 autochthonous human cases, 2 of which were of the diffuse form, similar to that reported by Simpson et al. (1968). Díaz Nájera (1971) collected specimens of *L. diabolica* at Muzquiz, Coahuila, a site where one of the patients lived. This sand fly, the only species known to bite people in Texas and northern Mexico, is the suspected vector. The nonforested, relatively dry habitat in these areas differs greatly from classical *Leishmania mexicana* foci in tropical forests of southern Mexico, Central and South America.

Visceral leishmaniasis in dogs

MacVean et al. (1979) and Anderson et al. (1980) reported visceral leishmaniasis in hunting dogs (fox hounds) from a kennel at Edmond near Oklahoma City, Oklahoma. The disease continues to be transmitted to dogs at this site, but its origin remains unknown. The causative agent is similar to *Leishmania donovani infantum*, a parasite causing canine and human visceral leishmaniasis in the Old World (Decker-Jackson and Tang 1982, Kocan et al. 1983), but Kreutzer et al. (1983) identified the parasite as *Leishmania mexicana*. MacVean et al. (1979) captured specimens of *Lutzomyia* near the kennel in the forest. We provisionally identified one of the males as *L. vexator*, on the basis of a drawing of the gonostylus kindly given to us by R. A. Ward, Walter Reed Army Institute of Research. The vector of leishmaniasis at this site remains unknown. *Leishmania*-infected dogs are occasionally imported into the United States and Canada from endemic areas (Maness 1981, Simpson et al. 1982) and there is recent serological evidence that coyotes from Texas may be infected with the disease (Grogl et al. 1984).

Rio Grande Virus in vertebrates

Rio Grande virus, family Bunyaviridae, genus *Phlebovirus* (Bishop et al. 1980), was named by Calisher et al. (1977) who recovered the virus from *Neotoma* woodrats near Brownsville, Texas and found serological evidence of infection in other vertebrates but not in people. *Lutzomyia anthophora* (Addis) is the suspected vector based on the habits of the species, its close association with woodrats, and its proven ability to vertically* transmit the virus in the laboratory (Endris et al. 1983).

Parasites of amphibians and reptiles

Ayala (1973) gave references to his earlier, detailed studies on amphibian and reptile parasites associated with phlebotomines, especially *L. vexator*, in central California. He showed that sand flies were suitable hosts for *Trypanosoma bufophlebotomi* Ayala of toads and *T. scelopori* Ayala and *T. gerrhonti* Ayala and McKay of lizards. A saurian malaria, *Plasmodium mexicanum* Thompson and Huff, developed to the sporozoite stage in sand flies and was infective when inoculated into *Sceloporus* fence lizards. Wild-caught sand flies also were found infected with hemogregarines (*Hepatozoon* sp. oocysts) that infected both lizards and snakes following experimental inoculations (Ayala 1970a).

METHODS AND MATERIALS

FIELD COLLECTIONS

The diversity of the sand fly fauna in a given area is best determined by using a combination of collection techniques. Such techniques were discussed by Lewis (1973, 1974) and Chaniotis (1978). In the present study, adult sand flies were routinely searched for in their diurnal resting sites, including tree trunks, under loose bark of dead standing trees, tree hollows, animal burrows and dens, rock crevices and other protected microhabitats. Specimens were captured with a simple tube aspirator, burrow traps and sticky paper traps. Endris et al. (1982) outlined procedures for handling such wild-caught sand flies for rearing purposes.

Flight traps and CDC light traps were used whenever possible (see Gressitt and Gressitt 1962, Sudia and Chamberlain 1962). They are effective devices for capturing most, but not all species. Limited attempts to recover immature

* Direct transfer of the virus from parent to its offspring.

stages from soil samples using a Berlese funnel were unsuccessful. As far as is known, immatures have not been found in nature in North America.

PRESERVATION AND SLIDE MOUNTING

It is recommended that adult sand flies be stored dry between layers of tissue paper in a pill box before they are mounted on slides because specimens preserved in alcohol are much more difficult to macerate, particularly those that have been stored in the preservative for a year or more. Following maceration in 10% NaOH, however, the flies can be stored indefinitely in 70% EOH. Methods for preparing slide mounts and identifying sand flies are outlined below following Young (1979).

1. Place whole sand fly in 10–25% sodium hydroxide (NaOH) at room temperature in a small crucible for 30–60 minutes.
2. Heat the NaOH to the boiling point on a hot plate, then immediately remove crucible from heat and allow to cool for 30 minutes.
3. Put macerated fly directly into a drop of 90% liquid phenol in the depression in a depression microscope slide (temporary slide mount) and identify with the aid of a compound microscope. A coverslip can be placed over the specimen if necessary. The internal spermathecae of female sand flies should then be clearly visible.
4. Preserve in 70% alcohol in a vial, or proceed to step 5 if a permanent slide mount is needed.
5. Place one drop of Canada balsam in the depression of another microscope slide. Mix 2–5 drops of phenol with it and put specimen or specimens (up to 12) in the mixture.
6. Allow the phenol to slowly evaporate at room temperature until the balsam-phenol mixture becomes somewhat viscous. The mixture has now infiltrated the body of the sand fly and the spermathecae and other structures should be normal in appearance.
7. Dissect the specimen by removing the head and wings. Lift these parts and the remainder of the specimen with a small needle and position them on a coverslip in small drops of the balsam-phenol mixture in which the fly was dissected. Orient the head so that the cibarium can be viewed as in the illustrations in this paper.
8. Place a small piece of glass, from a previously ground coverslip, in each corner of the coverslip holding the dissected sand fly. Allow to dry until the drops of mixture are completely hard.
9. Invert coverslip over a drop of Canada balsam placed in the middle of a clean microscope slide. Store the slide horizontally until dry.

MATERIAL EXAMINED

A number of colleagues generously supplied specimens, locality records (specimens not examined by the present authors), and other pertinent information. We gratefully acknowledge the help of F. S. Blanton, J. F. Butler, R. G. Endris, G. B. Fairchild, R. C. Johnson, W. L. Kramer and R. C. Wilkerson, all associated with the University of Florida at the time of this study; J. F. Reinert, R. W. Intermill, P. G. Lawyer and J. I. Glick, US Army medical entomologists; R. H. Roberts, ARS, USDA, Gainesville; W. W. Wirth, US Department of Agriculture; R. J. Brenner and S. Frommer, University of California, Riverside; Q. D. Wheeler, Cornell University; L. D. Hendricks and R. A. Ward, Walter Reed Army Institute of Research; Personnel of the US Army Health Services Command at various posts in the continental USA and C. G. Moore and D. B. Francy, Centers for Disease Control, Ft. Collins, Colorado.

Dr. V. F. Newhouse, Centers for Disease Control, deserves special recognition for allowing us to publish his records of North American sand flies identified earlier by G. B. Fairchild and W. J. Hanson but not confirmed by the present authors.

Holotypes of all new species will be deposited in the US National Museum of Natural History along with paratypes and other specimens. Paratypes also will be deposited in the collection at the University of Florida, and elsewhere as indicated in the text.

TAXONOMIC TREATMENT

Many of the species of *Lutzomyia* were originally described in the genus *Phlebotomus* Rondani and Berté (= *Flebotomus*)), a name now applied strictly to the medically-important group of Old World phlebotomines (Theodor 1965). The subgeneric and species group classification of *Lutzomyia* follows that of Lewis et al. (1977) and Martins et al. (1978). The morphological terminology agrees with that used by Quate and Vockeroth (1981). Thus, gonostylus and gonocoxite are substituted for style and coxite, respectively. The ejaculatory apodeme and sperm pump together refer to the familiar term, genital pump, that is used by many students of Phlebotominae. For quick reference, the structures shown in Fig. 2 are labelled. The immature stages are not treated in this review because of a lack of material, but references to them are given. Well known species are not redescribed because adequate descriptions are available in other papers cited here in each species account.

SYSTEMATIC ACCOUNT

CHECK LIST OF NORTH AMERICAN PHLEBOTOMINAE

Genus *Lutzomyia* França 1924

Subgenus *Lutzomyia* França 1924

1. *L. cruciata* (Coquillett 1907)
2. *L. diabolica* (Hall 1936)

Species Group *migonei* Theodor 1965

3. *L. xerophila* Young, Brenner and Wargo 1983

Subgenus *Coromyia* Barretto 1962 (= Species Group *vespertilionis* Theodor 1965)

4. *L. aquilonia* (Fairchild and Harwood 1961)

Subgenus *Dampfomyia* Addis 1945

5. *L. anthophora* (Addis 1945)

Subgenus *Psathyromyia* Barretto 1962 (= Species Group *shannoni* Theodor 1965)

6. *L. shannoni* (Dyar 1929)
7. *L. tanyopsis* Young and Perkins n.sp.

Species Group *aragaoi* Theodor 1965

8. *L. texana* (Dampf 1938)

Subgenus *Helcocyrtomyia* Barretto 1962 (= Species Group *vexator* Theodor 1965)

9. *L. apache* Young and Perkins n.sp.
10. *L. oppidana* (Dampf 1944)
11. *L. stewarti* (Mangabeira and Galindo 1944)
12. *L. vexator* (Coquillett 1907)
    = *L. vexator occidentis* Fairchild and Hertig 1957 NEW STATUS

Subgenus *Micropygomyia* Barretto 1962 (= Species Group *cayennensis* Theodor 1965)

13. *L. californica* (Fairchild and Hertig 1957)
14. *L. cubensis* (Fairchild and Trapido 1950)

KEY TO THE NORTH AMERICAN SPECIES OF *LUTZOMYIA*

It is usually necessary to prepare slide mounts of sand flies in order to study the structures of taxonomic importance. Once the local fauna becomes known, however, it is often possible to identify specimens simply by using color characters, relative lengths of the legs, body, wings and other features which can be seen without the aid of a microscope. In general appearance most sand flies resemble the one shown in Fig. 1. Sexual dimorphism is marked. The male has conspicuous external terminalia, its abdomen is relatively slender and it is usually smaller than the conspecific female.

*Males**

1. Gonostylus of genitalia with 5 large spines, subterminal seta absent. Gonocoxite with a basal tuft or group of persistent setae 2
   Gonostylus with 2–4 large spines; subterminal seta and gonocoxal tuft present or absent 6
2. Cibarium with a row of 8 or more short horizontal teeth. Gonocoxal tuft of 10–15 setae . . . . . *L. californica* (Fig. 14)
   Cibarium without a row of horizontal teeth. Gonocoxal tuft of 4–7 setae . . 3
3. Paramere with dorsal setae restricted to apical third. Aedeagal filaments beyond apical inflated portion shorter than inflated portion . . . *L. oppidana* (Fig. 11)
   Paramere with dorsal setae covering at least apical half. Aedeagal filaments beyond apical inflated portion longer than inflated portion 4
4. Lateral lobe barely extending beyond end of paramere. Gonostylus with paired basal spines inserted at same level, subterminal spine close to terminal pair. Antennal ascoids on flagellomere II shorter than one third the length of flagellomere . . . . *L. stewarti* (Fig. 12)
   Lateral lobe extending well beyond end of paramere. Gonostylus with basal spines placed at different levels, subterminal spine more or less isolated from terminal pair. Antennal ascoids on flagellomere II longer than half the length of flagellomere 5
5. Antennal ascoids on flagellomere II extending beyond end of flagellomere . . . . *L. apache* n.sp. (Fig. 10)
   Antennal ascoids on flagellomere II not reaching end of flagellomere . . . . *L. vexator* (Fig. 13)
6. Gonostylus with 2 or 3 strong spines, subterminal seta present. Paramere with an acute ventral projection . . . 7
   Gonostylus with 4 strong spines, subterminal seta absent. Paramere with or without an acute ventral projection . . 8
7. Paramere with a dorsal arm bearing recurved setae at enlarged end. Gonostylus with proximal spine much thinner than others. Gonocoxal tuft absent. Lateral lobe not inflated. Aedeagal filaments shorter than 3 times length of sperm pump and ejaculatory apodeme . . . *L. anthophora* (Fig. 6)
   Paramere simple, without a dorsal arm. Gonostylus with proximal spine about the same size as other. Gonocoxite with basal tufts of setae. Lateral lobe inflated. Aedeagal filaments longer than 3 times length of sperm pump and ejaculatory apodeme . . . *L. aquilonia* (Fig. 5)
8. Antennal ascoids with distinct proximal spurs. Palpomere 5 short (Figs. 8, 9) . . . 9
   Antennal ascoids simple, without proximal spurs. Palpomere 5 longer (Figs. 4, 12) 10
9. Gonocoxite with numerous setae at middle and beyond. Lateral lobe longer than coxite. Antennal ascoids extending beyond ends of flagello-

* The male of *Lutzomyia tanyopsis* n.sp. is not shown.

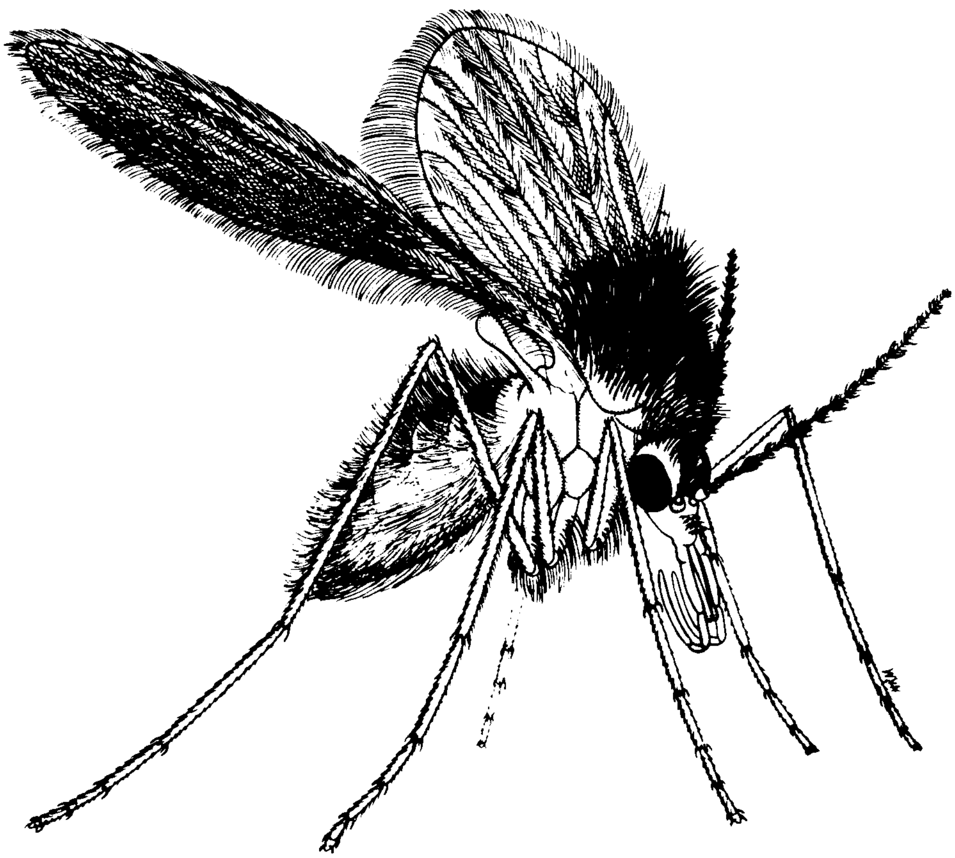

Fig. 1. A female phlebotomine sand fly.

meres, proximal spurs relatively short as shown .... *L. texana* (Fig. 9)
Gonocoxite without persistent setae. Lateral lobe shorter than gonocoxite. Antennal ascoids not reaching ends of flagellomeres, proximal spurs much longer .... *L. shannoni* (Fig. 7)

10. Gonocoxite lacking persistent setae .... *L. cubensis* (Fig. 15)
Gonocoxite with 1 or more persistent setae 11

11. Aedeagal filaments shorter than twice length of ejaculatory apodeme and sperm pump; filament tips enlarged, each with 5 inner teeth. Gonocoxite with 1–2 setae at inner base. Whole insect pale .... *L. xerophila* (Fig. 4)
Aedeagal filaments over twice as long as ejaculatory apodeme and sperm pump; filament tips simple, without teeth. Gonocoxite tuft of 4 or more setae inserted on a circular base. Scutum dark 12

12. Pleuron much paler than scutum. Flagellomere I extending to level of palpomere 3. Gonocoxite with 12–16 relatively thick persistent setae. Wings grayish .... *L. cruciata* (Fig. 2*)
Pleuron as dark as scutum. Flagellomere I relatively short, extending to level of palpomere 2. Gonocoxite with fewer than 10 slender persistent setae. Wings yellowish .... *L. diabolica* (Fig. 3)

Females

1. Pharynx armed with posterior spines .... *L. cubensis* (Fig. 15)
Pharynx unarmed 2

2. Antennal ascoids with proximal spurs 3
Antennal ascoids simple, without proximal spurs 5

3. Cibarium with 8 or more horizontal teeth. Antennal ascoids with short proximal spurs as shown in Fig. 9. Spermathecae large and spherical without annulations .... *L. texana* (Fig. 9)
Cibarium with 4 horizontal teeth. Antennal ascoids with long proximal spur ending near base of flagellomeres. Spermathecae tubular or spherical with basal annulations 4

4. Spermathecae tubular, much longer than wide, without annulations; common sperm duct longer than individual duct. Head width and height subequal; eyes relatively large as shown in Fig. 7 .... *L. shannoni* (Fig. 7)
Spermathecae spherical, as long as wide, with basal annulations; common sperm duct shorter than individual duct. Head much longer than wide; eyes relatively small ..... *L. tanyopsis* (Fig. 8)

5. Cibarium with a comb-like row of 14 or more horizontal teeth. .. *L. californica* (Fig. 14)
Cibarium with 2–10 horizontal teeth 6

6. Cibarium with 2 broadly rounded or blade-like horizontal teeth and with numerous lateral teeth 7
Cibarium with 4–10 pointed horizontal teeth; lateral teeth inconspicuous or absent 8

7. Cibarium with 2 rounded horizontal teeth, each with 1 or 2 short spines. Spermathecae wrinkled, oval without bubble-like evaginations . *L. aquilonia* (Fig. 5)
Cibarium with 2 blade-like horizontal teeth. Spermathecae modified with numerous bubble-like evaginations .... *L. anthophora* (Fig. 6)

8. Individual sperm ducts shorter than stem of genital fork. Spermathecae with an outer envelope arising from base of spermathecae .... *L. xerophila* (Fig. 4)
Individual sperm ducts longer than stem of genital fork. Spermathecae differently shaped and lacking an outer envelope 9

9. Spermathecae spherical, spermathecal width the same as, or greater than, width of common sperm duct; with distinct annulations at base 10
Spermathecae button-like, width less than width of common sperm duct, without basal annulations, but transverse striations or excrescences may be present on individual duct .. 12

10. Labrum shorter than combined length of flagellomeres II + III and shorter than twice the length of palpomere 5. Cibarium with only 4–6 very small vertical teeth .... *L. stewarti* (Fig. 12)
Labrum subequal to, or longer than, combined lengths of flagellomeres II + III and longer than twice the length of palpomere 5. Cibarium with 10 or more vertical teeth 11

11. Tergite nine with 2 heavily sclerotized, anterolateral papillate lobes. Labrum shorter than flagellomere I. Cibarium with small vertical teeth in 2–3 irregular rows; pigment patch shaped as shown in Fig. 2. Individual sperm ducts at least 6 times length of spermatheca. Scutum dark, pleuron pale. Wings grayish .... *L. cruciata* (Fig. 2*)
Tergite nine without papillate lobes. Labrum longer than flagellomere I. Cibarium with 4–7 large vertical teeth in a single row; pigment patch otherwise, relatively broad and darker at expanded base. Individual sperm ducts about 4 times length of spermatheca. Scutum and pleuron pale. Wings yellowish .... *L. diabolica* (Fig. 3)

* Applies to specimens from the USA but is not always valid for Mexican and Central American specimens

12. Individual sperm ducts relatively short, not over 3 times length of stem of genital fork .... *L. vexator* (Fig. 8F & 13)
Individual sperm ducts over 5 times length of stem of genital fork 13
13. Antennal ascoids of flagellomere II extending beyond end of flagellomere. Individual sperm ducts relatively slender as shown in Fig. 10 ... *L. apache* (Fig. 10)
Antennal ascoids of flagellomere II not extending to end of flagellomere. Individual sperm ducts wider, as shown in Fig. 11 .... *L. oppidana* (Fig. 11)

Subgenus *Lutzomyia* França

1. *Lutzomyia (Lutzomyia) cruciata* Coquillett, Fig. 2.

Flebotomus cruciatus Coquillett 1907:102 (♀, Trece Aguas, Alta Vera Paz, Guatemala).

Phlebotomus cruciatus: Barretto 1947:194 (full refs.). Fairchild and Hertig 1948a:247 (♀ tax., refs.). Biagi and Biagi 1953a:315 (dist., habits, tax., Mexico). Vargas and Díaz Nájera 1953:310 (dist., Mexico). Fairchild and Hertig 1953:377 (♂, ♀, tax.). Fairchild 1955:192 (listed). Rosabal 1954:22 (tax., Costa Rica). Fairchild and Hertig 1959:12 (dist., Central America). Rosabal and Trejos 1964:167 (El Salvador records); 1965:222 (El Salvador), Biagi et al. 1965:267 (coll. data, Mexico). Biagi 1966:368 (keyed).

Brumptomyia cruciata: Lewis 1965:375 (internal structures, Belize)

Lutzomyia cruciata: Theodor 1965:182 (classification). Williams 1966:365 (transmission of *Leishmania mexicana*, Belize). Disney 1968:267 (as senior synonym of *L. diabolica*). Williams 1970:33 (summary of coll. data, Belize). Lewis 1971:538 (listed, Florida). Young 1972:63 (Florida records). Forattini 1973:243 (tax., dist., refs.). Eads 1978:538 (listed). Martins et al. 1978:121 (dist. records). Young et al. 1981:446 (larval food). Endris et al. 1982:401 (rearing data). Zeledón et al. 1982:276 (dist. records, Honduras).

Known distribution: Panama to SE Mexico and USA. *Florida* (Alachua and St. Johns counties); *Georgia* (Charlton Co.), Fig. 18.

Material examined: USA. *Florida*—1 ♀ (Alachua Co.), San Felasco Hammock, 23-V-1960, light trap, F. Blanton. 1 ♀, same data but VI-1964. 5 ♀, same data but 14-V to 30-X-1967. 1 ♀, same data but 14-V-1970, flight trap, D. Young. 1 ♀, same data, but 9-VIII-1971, light trap, D. Young. 8 ♀, same data, but 11-IV-1975 and 25-IV to 23-V-1977, flight trap, H. Greenbaum and G. Fairchild. 1 ♀ (Alachua Co.), Gainesville, 2 2-VII-1980, tree trunk, D. Young. 1 ♂, 2 ♀ (and 33 ♂, 44 ♀ progeny) (Alachua Co.), San Felasco Hammock, 16-IX-1980, tree trunk, P. Perkins. 19 ♀ (Alachua Co.), Gainesville, VII to VIII-1981, light traps (20 trap nights), D. Young. 1 ♀ (St. Johns Co.), Anastasia State Park, 12-V-1979, flight trap, D. Young. *Georgia*—1 ♀ (Charlton Co.), Billy's Island, Okefenokee Swamp, 16-IX-1965, light trap, V. Newhouse. Mexico. *Campeche*—1 ♀, Matamoros, 20-I-1944, L. Vargas. *Chiapas*—1 ♀, La Granja, VII-1930, no other data. 8 ♀, Tapachula, XII-1941, M. Macias. 1 ♀, Palenque, 31-III-1951. Shannon trap, G. Fairchild and R. Hartmann. 1 ♀, Finca Guadalupe, Zaju, 24-X-1940, Diaz Nájera. 1 ♀, Santa Maria, Cintalapa, II-IV-1951, G. Fairchild and R. Hartmann. *Hidalgo*—1 ♀, near Santa Ana, 15-IX-1955, P. Galindo and H. Trapido. *Quintana Roo*—1 ♀, Centote Azul, 8-X-1954, P. Galindo and H. Trapido. *San Luis Potosi*—2 ♀, Tamazunchale, VI-1942, M. Macias. *Tabasco*—5 ♀, Arroyo Expangale, VII to 7 VIII-1941, M. Macias. 23 ♀, Teapa, IV to XI-1953, R. Alegria and Figueroa.

Guatemala—*Escuintla*—3 ♂, N of Escuintla. 17-V-1945, tree buttresses, G. Fairchild. *Peten*—1 ♀, Tikal, 17-IV-1960, biting man, E. McConnell. 2 ♂, 22 km S of San Francisco, 7-V-1980, tree buttresses, C. Porter. 1 ♀, Finca el Zapote, 8-IX-1980, light trap, C. Porter. *Solola*—1 ♀, Finca St. Basilio, no date, Morales. Belize, 1 ♀, 56 km SW Belize City on road to Roaring Creek, 6-X-1955, biting man, P. Galindo and H. Trapido. El Salvador. *San Miguel*—4 ♂, San Jorge, IX-1966, 1967 and X-1967, light traps, F. Blanton. 1 ♂ Moncagua, 9-XII-1966, light trap, F. Blanton. *San Vicente*—1 ♀, Santo Domingo, XI-1966, light trap, F. Blanton. *Sansonate*—1 ♀, Armenia, I-1967, light trap, F. Blanton.

Honduras, *Atlántica*—3 ♂, 40 ♀, Tela Lancetilla Valley, 12-XI-1953 and 24-III-1954 to 7-VI-1954, light traps, biting man and tree trunks, W. Hils. *Francisco Morazan*—2 ♂, 1 ♀, Zamorano, III and 8-VI-1966, light traps, J. Matta and F. Blanton. Nicaragua. *Carazo*—2 ♂, 5 ♀, Guapinolar, VIII-X-1953, A. Adames. *Chontales*—3 ♂, 2 ♀, Villa Samoza, V and IX-1953, P. Galindo and H. Trapido. *Managua*—1 ♀, Casa Colorado near Managua, XII-1953, biting man, J. Boshell.

Costa Rica, *Cartago*—1 ♀, Turrialba, 11-II-1952, Shannon trap, R. Rosabal and M. Hertig. 1 ♀, Turrialba, on horse, 2-V-1961, R. Rosabal and M. Hertig. *Guanacaste*—1 ♂, Lagartos, at Guanacaste-Puntarenas border, 13-XII-1951, tree buttress, R. Rosabal.

Panama. *Boca del Toro*—2 ♂, 2 ♀, Almirante, 19-22-VI-1950, tree buttresses, R. Hartmann.

1 ♂, 12 ♀, Almirante, 19-VI to 18-XI-1951, tree buttresses and Shannon trap, A. Quiñones. 1 ♀, Almirante, 6-II-1952, biting man, A. Quiñones. Canal Zone—1♀, Rio de Medio at Gatun River, 13-X-1949, hollow tree, R. Hartmann. 2 ♂, Madden Forest, 10-VIII-1951 and 25-II-1952, light trap and tree buttress, M. Hertig, R. Rosabal and R. Hartmann.

DISCUSSION: Both sexes of *L. cruciata* from North America have relatively small eyes (Fig. 2) when compared to those of *L. cruciata* from the neotropics and *L. diabolica* (Fig. 3). This character state and the slightly thicker setae of the coxite tuft of Florida males support the idea that the nearctic population of *L. cruciata* is an isolated one that may have been derived from neotropical stock that spread through the Gulf of Mexico coastal plain into Florida and Georgia. It is also possible that individuals entered Florida by crossing the Gulf of Mexico, possibly with the aid of hurricane winds, but this hypothesis seems less likely. *Lutzomyia cruciata* is not known to occur in the West Indies.

Existing populations of *L. cruciata* in Florida and Georgia are geographically isolated from those in southeastern Mexico and Central America and it does not appear that this species presently occurs in states west of Florida or in northern Mexico. *Lutzomyia cruciata* and *L. diabolica* have allopatric distributions in the USA (Fig. 18) but the extent of their geographic distributions in Mexico has not been determined. *Lutzomyia cruciata* occurs mainly in secondary and primary hardwood forests, but *L. diabolica* has been found in dry, sometimes treeless localities (e.g., area of Comstock, Texas).

All of the *L. cruciata* females from Florida and Georgia have a papillate lobe on each side of the 9th abdominal tergite (Fig. 2D). This modification is absent in females of *L. diabolica*, *L. gomezi* (Nitz.) and some *L. cruciata* females from Mexico, Guatemala, Honduras and Nicaragua.

In Belize, Lewis (1965) observed that 96% of *L. cruciata* females dissected were parous, thus indicating that the species is normally autogenous. Perkins (1982) established a laboratory colony of *L. cruciata* from a gravid female collected near Gainesville (Alachua Co.), Florida. All females from the F_1 through F_{26} generations were autogenous and deposited 35.8 ± 13.5 eggs (range 1–60), 3–10 days following emergence. Other rearing data based on this colony are given by Perkins (1982).

Eighteen females of *L. cruciata* were collected in CDC light traps near Gainesville (Sugarfoot Hammock) in August and September, 1980 (8 trap nights). Five of them, held in a 120 ml feeding chamber (Endris et al. 1982), took blood meals on the arm of a human volunteer during a 1–3 minute period. Their bites were quite painful when compared to those of *L. shannoni*. All 18 females died without depositing eggs and most lived 2.9 ± 2.4 days after capture. The specimens that took human blood meals survived from 6 to 8 days but none had mature eggs when dissected. Lab-reared females were given the opportunity to feed on man on numerous occasions but none, including those that had oviposited 1–7 days previously, did so.

There is little evidence indicating that *L. cruciata* is a natural vector of cutaneous leishmaniasis (*Leishmania mexicana*) in Belize (Williams 1970). Williams (1966), however, demonstrated that *Leishmania mexicana* multiplies and develops in *L. cruciata* females and that experimentally-infected flies transmitted the parasite by bite to a human volunteer.

2. *Lutzomyia (Lutzomyia) diabolica* (Hall), Fig. 3.

Phlebotomus sp.: Parman 1919:211 (biting man, Texas)

Phlebotomus diabolicus Hall 1936:28 (♂, ♀, Uvalde, Uvalde Co., Texas). Lindquist 1936:29 (life cycle, biol., descript. immatures, Sonora, Sutton Co., Texas record). Dampf 1938:121 (mention). Rozeboom 1944:274 (listed). Addis 1945a:328 (keyed); 1945b:119 (tax.). Packchanian 1946:37 (listed). Barretto 1947:197 (refs.). Fairchild and Hertig 1948a:25 (compared to *L. gomezi* and *L. cruciata*). Thurman et al. 1949:199 (listed). Vargas and Díaz and Nájera 1953b:311 (Mexico). Fairchild and Hertig 1953b:375 (♂, ♀, keyed, redescript., refs.). Quate 1955:242 (keyed, refs.). Fairchild and Hertig 1959:123 (dist., Mexico). Lewis and Garnham 1959:86 (mention). Eads et al. 1965:251 (listed). Quate 1965:92 (listed). Easton et al. 1968:466 (Brackettvile area, Kinney Co., Texas).

Lutzomyia diabolica: Theodor 1965:182 (listed). Disney 1968:267 (as junior synonym of *L. cruciata*). Díaz Nájera 1971:62 (Coahuila State, Mexico) Forattini 1973:252 (Fig., tax.). Endris et al. 1982:401 (reared in laboratory).

Lutzomyia cruciata, not *cruciata* Coquillett 1907: Rosabal and Miller 1970:180 (♂, ♀ keyed). Martins and Morales Farias 1972:365 (in part, dist.). Young 1972:61 (in part, Texas records). Calisher et al. 1977:1000 (Brownsville, Texas). Eads 1978:539 (in part, listed, refs.). Martins et al. 1978:121 (dist., Texas and Mexico). Chaniotis 1978:19 (mention).

Lutzomyia cruciata diabolica: Lewis 1975:509 (mouthpart morphol.)

KNOWN DISTRIBUTION: SW Mexico to USA *Texas* (Atacosa, Bexar, Cameron, Comal, Edwards, Gillespie, Hale, Kinney, Llano, Medina,

Nueces, Sutton, Tom Green, Travis, Uvalde and Val Verde counties), Fig. 18.

Material examined: USA. *Texas*—1 ♀ (Atascosa Co.), Poteet (USNM). 9 ♀ (Bexar Co.), Camp Bullis, 16-VI-1965, light traps, D. Young. 1 ♀ (Cameron Co.), Laguna Atascosa Wildlife Refuge, 26-IV-1965, light trap, R. Eads. 1 ♀ (Comal Co.), Canyon Lake, 14-X-1971, biting man, 1400 hrs., R. Wilkerson. 20 ♀ (Edwards Co.), 6 km N of Barksdale, V-1982, at light, P. Lawyer et al. 5 ♂, 5 ♀ (Gillespie Co.), Fredericksburg, VII to IX-1968 and V-1969, light traps, H. Borchers. 1 ♀ (Hale Co.), Plainview, VII-1971, light trap, VEE Survey Team. 2 ♀ (Llano Co.), Lake Buchanan, 17-VII-1966, light trap, D. Young. 1 ♂, 6 ♀ (Medina Co.), D'Hanis, 28-29-IX-1983, light traps, P. Lawyer. 1 ♀ (Nueces Co.), Corpus Christi, biting man (USNM). 1 ♀ (Travis Co.), Austin, (USNM). 2 ♂, 14 ♀ (Uvalde Co.), Garner State Park, 17-V-1965, D. Young. 500+ ♀, same data but 2-VI to 28 VI-1982, in latrines, biting man, flight and light traps, P. Lawyer et al. 2 ♂, 20 ♀ (Val Verde Co.), Comstock, VIII-1971, J. Reinert, VEE Survey Team. Mexico. *Chihuahua*—1 ♂, 1 ♀, 4.5 km SE Hidalgo del Parral, 22-VIII-1975, UV light trap, W. Sumlin (UCR). *Morelos*—3 ♂, 3 ♀, Miacatlán, IV-V-1980, flight trap, M. Camino.

Additional records: *Texas*—2 ♀ (Tom Green Co.), San Angelo Reservoir, 11-VI-1968, light trap, V. Newhouse.

Discussion: Addis (1945b) redescribed *L. diabolica* from topotypic specimens, noting that the female had 6 horizontal teeth in the cibarium. Fairchild and Hertig (1948a, 1953b) distinguished the male of this species from that of *L. cruciata* by its more slender ejaculatory apodeme with its cup-shaped proximal end and by the presence of fewer and more slender setae of the gonocoxal tuft. Differences between the females of *L. diabolica* and *L. cruciata* were mainly those related to the size and number of vertical teeth in the cibaria, the former species having only 8–10 heavy blunt teeth; the latter with more numerous, smaller vertical teeth. Dorsolateral, papillate lobes of the ninth abdominal tergite were observed in females of both species but were less developed in *L. diabolica*. Only 4 females of *L. diabolica* were available to these authors for study.

Disney (1968) studied structural variation in numerous males and females of *L. cruciata* from Belize, concluding that it was not possible to separate this species from *L. diabolica* based on available criteria. He observed that the proximal end of the ejaculatory apodeme was of the "*cruciata* form" in 80% of the Belize males examined. The basal gonocoxal setae varied in number from 8 to 23 ($\bar{x}$ = 16), and the number of vertical teeth in the female cibarium ranged from 8 to 29 ($\bar{x}$ = 16). Other features, including the spermathecae, chitinous arch of the cibarium and number of episternal setae, were studied. From his analysis, he concluded that variation among his material was infraspecific and that the name *diabolica* should be "suppressed, at least as a specific epithet."

In the USA and Meixco, however, there are 2 distinct forms that can be identified as *L. diabolica* and *L. cruciata* and which are easily distinguished by the characters given in the keys. Disney (1968) was correct in assigning his Belize specimens to the latter species; *L. diabolica* is not known to occur there or in other countries south of Mexico but there is a good possibility that populations exist in western Guatemala.

Lutzomyia diabolica is very dark; the pleuron of each sex is as dark as the scutum, unlike *L. cruciata* which has a pale pleuron. There seems to be no single, consistent structural difference between the 2 species although the curious papillate lobes of the 9th tergite are present in all *L. cruciata* females from the USA and in 83% (n = 81) of *L. cruciata* females from Mexico and Central America. Females of *L. diabolica* lack such lobes.

The ratio of the length of flagellomere 1 to the head height (flag. 1/head height) of *L. diabolica* females ranges from 0.45 to 0.61 (n = 31, $\bar{x}$ = 0.53, SD = 0.032); that for males ranges from 0.52 to 0.63 (n = 5, $\bar{x}$ = 0.568, SD = 0.041). In females of *L. cruciata* females from Mexico and Central America (n = 73), this ratio varies from 0.61 to 0.88 ($\bar{x}$ = 0.732, SD = 0.056); that for *L. cruciata* males ranges from 0.66 to 0.86 (n = 25, $\bar{x}$ = 0.756, SD = 0.048). The number of horizontal teeth in the female cibarium of both species ranges from 4 to 7.

Females of *L. diabolica* are agressive man-biters, attacking mostly at night but occasionally in the daytime as well. The natural resting and breeding sites of this species are unknown. At Garner State Park, Uvalde Co., Texas, about 1500 specimens were collected from late May to early June 1983 on the inside walls of latrines. The new record of this species from Plainview, Texas, represents the northernmost limit of its known geographic range but few attempts have been made to collect this species in northern Texas, New Mexico or Oklahoma.

Lindquist (1936) and Endris et al. (1982) reared *L. diabolica* in the laboratory; the former author providing information on its life cycle and describing the immature stages. Females are anautogenous. Additional field studies of this species are recommended in view of its man-biting habits and possible role as a vector

of cutaneous leishmaniasis in Texas and northern Mexico.

SPECIES GROUP *MIGONEI* THEODOR

3. *Lutzomyia xerophila* Young, Brenner and Wargo, Fig. 4.

Lutzomyia xerophila Young, Brenner and Wargo 1983:313 (♂, ♀, Cahuilla Hills, Palm Desert, Riverside Co., California).

KNOWN DISTRIBUTION: USA. *California* (Riverside Co.), Fig. 19.

MATERIAL EXAMINED: USA. *California*—5 ♂, 150 ♀ (holotype, allotype and paratypes) type-locality, 12-IX-1980; 21-V-1981 to 4-X-1981, CO_2 traps, M. Wargo. 2 ♂, 24 ♀ (Riverside Co.), Rancho Mirage, Magnesia Springs Canyon, 16-VI-1981, CO_2 trap, R. Brenner.

DISCUSSION: *Lutzomyia xerophila* is provisionally placed in the *migonei* Group of *Lutzomyia* (Theodor 1965) on the basis of male and female structures, but the male lacks a subterminal seta on the gonostylus unlike other males in the group. This pale species is easily recognized by the characteristic spermathecae of the female and by the male terminalia. Intraspecific variation of the cibarial armature of females is shown in Fig. 4.

Specimens have been taken only in CO_2 traps in the Colorado Desert of Riverside Co., California from May to November. Nothing is known about the feeding habits, diurnal resting sites or biology of this recently described species.

SUBGENUS *COROMYIA* BARRETTO

4. *Lutzomyia (Coromyia) aquilonia* (Fairchild and Harwood), Fig. 5.

Phlebotomus aquilonius Fairchild and Harwood 1961:244 (♂, ♀, Columbia Wildlife Refuge, Othello, Adams Co., Washington). Eads et al. 1965:25 (listed). Harwood 1965:1 (locality records, Whitman Co., Washington and Southern Alberta, British Columbia, Canada). Quate 1965:91 (listed). Shemanchuk et al. 1978:1355 (seasonal dist., in marmot burrows, Police Coulee, Alberta, Canada).

Lutzomyia aquilonia: Theodor 1965:184 (listed). Rosabal and Miller 1970:180 (♂, ♀ keyed). Forattini 1971:99 (listed). Martins and Morales-Farias 1972:365 (listed). Forattini 1973:212 (keyed, brief tax. discussion). Chaniotis 1978:19 (listed) Martins et al. 1978:92 (dist.). Eads 1978:539 (dist., refs.). Young, 1979:91 (listed).

KNOWN DISTRIBUTION: Canada. *Alberta* and *British Columbia*. USA. *Colorado* (Larimer Co.); *Washington* (Adams and Whitman counties), Fig. 19.

MATERIAL EXAMINED: USA. *Colorado*—11 ♂, 5 ♀ (Larimer Co.), approx. 400 m east of Horsetooth Reservoir, light and oil traps, 18-VIII and 8-IX-1983, C. Moore and B. Francy. *Washington*—3 ♂, 3 ♀ (holotype, allotype and paratypes) (Adams Co.), Columbia Wildlife Refuge, Othello, VII-VIII-1959, 1960, R. Harwood.

DISCUSSION: The small median seta shown on the male gonostylus (Fig. 5A) is absent in the other males of this species examined. The female sperm ducts are illustrated for the first time following immersion of the slide-mounted specimen in 90% liquid phenol for one week.

Shemanchuk et al. (1978) collected 106 specimens of *L. aquilonia* in the burrows of the marmot, *Marmota flaviventris nosophora* Howell, during a 3-year study in Southern Alberta. Flies were collected from August to October at one site, and in July and August at the Columbia Wildlife Refuge, Washington State (Fairchild and Harwood 1961). The specimens from Colorado were collected near nests of *Neotoma* woodrats located in rock crevices.

The feeding habits of this species remain unknown.

SUBGENUS *DAMPFOMYIA* ADDIS

5. *Lutzomyia (Dampfomyia) anthophora* (Addis), Fig. 6.

Phlebotomus anthophorus Addis 1945b:119 (♂, ♀, Uvalde, Uvalde Co., Texas); 1945c:319 (rearing data, descript. immature stages); 1945:328 (keyed). Packchanian 1946:37 (refs.). Barretto 1947:185 (refs.). Thurman et al. 1949:199 (listed). Vargas and Díaz Nájera 1953a:46 (Michoacan State, Mexico, compared to *L. dodgei*); 1953b:309 (Morelos State, Mexico). Quate 1955:242 (keyed, refs.). Fairchild and Hertig 1956:307 (dist., keyed, refs.); 1959:123 (Nuevo León State, Mexico). Forattini 1959:195 (rearing data from Addis 1945c). Chaniotis and Anderson 1964:27 (mention). Quate 1965:91 (listed). Eads et al. 1965:251 (listed). Easton et al. 1968:466 (Presidio and Kinney Co., Texas records).

Lutzomyia anthophora: Barretto 1962:95 (listed). Theodor 1965:193 (listed). Rosabal and Miller 1970:180 (♂, ♀ keyed). Williams 1970:332 (mention). Forattini 1971:100 (listed). Young 1972:61 (in woodrat nests, Texas). Martins and Morales-Farias 1972:365 (listed). Forattini 1973:122 (tax.). Calisher et al. 1977:1000 (Cameron Co., Texas, possible vector of Rio Grande Virus). Martins et al. 1978:57 (dist.). Eads 1978:539 (dist., refs.). Chaniotis 1978:19 (mention). Young et al. 1981:446 (larval food). Endris et al. 1982:401 (rearing data,

lab. hosts). Endris et al. 1983:862 (transovarial transmission of Rio Grande Virus).

KNOWN DISTRIBUTION: Mexico to USA. *Texas* (Bexar, Cameron, Kinney, Medina, Presidio, San Patricio, Uvalde and Val Verde counties), Fig. 19.

MATERIAL EXAMINED: USA. *Texas*—1 ♂, 3 ♀ (Bexar Co.), San Antonio, Lackland AFB, 20-XI-1965, D. Young and C. Parrish. 3 ♀ (Cameron Co.). Laguna Atascosa Wildlife Refuge, 26-IV-1965, light trap, R. Eads (USNM). 1 ♂, 1 ♀ (Cameron Co.), 12 km E of Brownsville, i-I-1966, *Neotoma* nest, D. Young. 39 ♂, 8 ♀, same data but V-1975. 2 ♀ (Medina Co.), D'Hanis, 29-IX-1983, *Neotoma* nest, P. Lawyer. 2 ♀ (San Patricio Co.), 12 km E of Sinton, 10-VI-1966, *Neotoma* nest, D. Young. 4 ♀ (Uvalde Co.), Uvalde, 28-IX-1944, on rabbits, H. Brundrett (USNM). 1 ♀ (Val Verde Co.), Del Rio, IX-1963, light trap, R. Eads. Mexico. *Morelos*—11 ♂, Miacatlán, 27-IV to 5-V-1980, flight trap, M. Camino. *Nuevo León*—1 ♂, 32 km NW of Monterrey, 9-IX-1955, rock crevice, P. Galindo and H. Trapido.

DISCUSSION. In addition to the material noted above hundreds of *L. anthophora* collected in *Neotoma* woodrat dens near Brownsville, Texas in 1966, 1975 and 1980 were examined.

The spermathecae with the unusual bubble-like evaginations (Fig. 6I) and the distinctive male terminalia (Fig. 6A) readily distinguish *L. anthophora* from other species in North America. It is currently impossible to separate females of this species from those of *L. dodgei* (Vargas and Díaz Nájera) from SW Mexico where they are sympatric. Vargas and Díaz Nájera (1953a) distinguished these females by differences in the cibarial armatures and spermathecae, characters that were found in this study to be unreliable for species identification.

Fairchild and Hertig (1956) believed that the female of *L. dodgei* described by Vargas and Díaz Nájera (1953a) actually was a female of *L. anthophora*. This conclusion was erroneously based on their study of specimens that were later named *L. atalupai* by De León (1971), not *L. dodgei* (Porter and Young, unpublished observations). Thus, *L. anthophora* and *L. dodgei* will key out together in couplet 2 of Fairchild and Hertig's 1956 key to the species of *Dampfomyia*.

Addis (1945b) reported *L. anthophora* females feeding on domestic rabbits at Uvalde, Texas. No specimens were taken on other hosts or from suspected resting sites. Young (1972) found both sexes of this species in the dens of woodrats, *Neotoma micropygus* Baird, and concluded that adults were present throughout the year in South Texas. Subsequently, one of us (D.G.Y.) collected more than 600 specimens of *L. anthophora* from a single woodrat den, east of Brownsville, Texas, on 31 May 1975. It was estimated that an equal number escaped capture. At that time, and in 1980, other specimens were also discovered resting in or near small rodent nests located underneath discarded lumber and cardboard. From these field associations and on laboratory observations, it is suspected that *L. anthophora* females feed primarily on small mammals; suitable laboratory hosts include hamster (Addis 1945c), white mouse, squirrel *(Sciurus)*, calf, rabbit, opossum *(Didelphis)* and domestic pig (Endris et al. 1982). Females feed to repletion and are gonotrophically concordant; 33 females (14%) out of 228 females that survived first oviposition in the laboratory took a second blood meal. Seven of these fed a third time and one took a partial fourth bloodmeal before dying (Endris and Young, unpublished observations).

Addis (1945c) and Endris (1982) studied the life cycle of *L. anthophora* in the laboratory. The former author used a rabbit blood-garden soil mixture for larval food; the latter used a composted rabbit feces-laboratory chow mixture (Young et al. 1981). At 28°C, the mean time from egg deposition to adult emergence was 49 days (n = 72, Addis 1945c) but was only 39 days in the later study (n = 41) (Endris 1982). These conflicting results apparently were due to differences in the larval diets. Addis (1945c) described the immature stages from reared material.

Endris et al. (1983) experimentally infected *L. anthophora* females with Rio Grande virus that was subsequently recovered from 54.8% of their F_1 progeny. This was the first virologically confirmed demonstration of vertical transmission of a *Phlebovirus* by sand flies. *Leishmania mexicana* (WRAIR strain 411 from Texas) was successfully transmitted to non-infected Syrian hamsters by the bites of experimentally infected, laboratory-reared *L. anthophora* (Endris and Young, unpublished observations).

SUBGENUS *PSATHYROMYIA* BARRETTO (= Species Group *shannoni*)

6. *Lutzomyia (Psathyromyia) shannoni* (Dyar), Fig. 7.

Phlebotomus shannoni Dyar 1929:121 (♂, Cano Saddle, Gatun Lake, Panama Canal Zone). Barretto 1947:222 (full refs., synonyms). Thurman et al. 1949:199 (biting man, Florida). Fairchild and Hertig 1950:523 (tax. review, Georgia record). Vargas and Díaz Nájera 1953b:312 (Mexican records). Snow 1955:515 (biting man, Tennessee). Quate 1955:242 (keyed, refs.).

Fairchild 1955:192 (classif.). Eads et al. 1965:21 (mention). Hanson 1968:78 (descript. larva, pupa).

Phlebotomus limai Fonseca 1935–1936:61 (♀, Sao Paulo, Brazil). Rozeboom 1944:274 (records from Alabama, Mississippi and North Carolina). Addis 1945a:328 (keyed). Packchanian 1946:38 (USA records).

Lutzomyia shannoni Barretto 1962:99 (as typespecies of subgenus *Psathyromyia*). Theodor 1965:189 (listed, figures male and female, classif.). Rosabal and Miller 1978:180 (keyed, coll. data Louisiana, man biting record). Young 1972:61 (coll. data, Florida). Forattini 1973:294 (tax. review). Zeledón and Alfaro 1973:416 (naturally infected with promastigotes). Lewis 1975:502 (mouthpart morphol.). Eads 1978:540 (dist., refs.). Chaniotis 1978:19 (mention). Martins et al. 1978:109 (dist., refs.). Young 1979:117 (refs., tax.). Young et al. 1981:446 (larval food). Endris et al. 1982:401 (rearing methods). Christensen and deVasquez 1982:243 (natural hosts, Panama).

KNOWN DISTRIBUTION: Argentina to USA. *Alabama* (Dale, Dallas, Lauderdale and Lee counties). *Arkansas* (Garland, Jefferson and Union counties). *Delaware* (New Castle Co.). *Florida* (Alachua, Collier, Columbia, Dade, Gadsden, Hernando, Jefferson, Levy, Liberty, Marion, St. Johns and Wakulla counties). *Georgia* (Charlton, Chatham, Clinch and Ware counties). *Louisiana* (Calcasiea, DeSoto, Jackson, Morehouse, Orleans, Rapides, St. Martin, St. Tammany and West Feliciana parishes). *Maryland* (Anne Arundel and Wicomico counties). *Mississippi* (Hancock and Hinds counties). *North Carolina* (Hoke, Jones, Onslow, Richmond and Duplin counties). *South Carolina* (Richmond Co.). *Tennessee* (Lake Co.), Fig. 19.

MATERIAL EXAMINED: USA. *Alabama*—1 ♂ (Dale Co.), Ft. Rucker, 23-IV to 11-VI-1981, light trap, pvnt. med. personnel, US Army. 4 ♂, 1 ♀ (Lee Co.), Chewacla State Park, 22-V; 5-X and 15-16-X-1976 light trap, J. Glick. *Arkansas*—1 ♂ (Garland Co.), Hot Springs, VIII-1971, light trap, J. Reinert. 1 ♂, 1 ♀ (Jefferson Co.), Pine Bluff, VIII-1971, light trap, J. Reinert. *Delaware*—1 ♀ (New Castle Co.), 7 km N of Wilmington, 1-VII-1981, tree trunk, R. Johnson. *Florida*—30 ♀ (Alachua Co.), San Felasco Hammock, 8-V-1969, light trap, D. Young. 1 ♀ (Alachua Co.), Gainesville, 6-XI-1975, tree trunk, D. Young. 230 ♂, 105 ♀ (Alachua Co.), same site, IV-1980 to X-1981, tree trunks and hollows, P. Perkins. 12 ♂, 92 ♀ (Alachua Co.), River Styx near Lochaloosa, 30-III to 2-IV-1982, CO_2 baited light tap, C. Atkinson. 8 ♂, 5 ♀ (Columbia Co.), Oleno State Park, IX/1973 and IX-1975, tree trunks, D. Young. 1 ♂, 1 ♀ (Gadsden Co.), 5 km SE Quincy, 23-24-VI-1980, tree trunk and light trap, D. Young and P. Perkins. 2 ♀ (Hernando Co.), Brooksville, 18-VI-1980, light trap, W. Kramer. 1 ♀ (Jefferson Co.), Monticello, 2-VI-1969, light trap, W. Whitcomb. 184 ♂, 60 ♀ (Levy Co.), Gulf Hammock, IV-1980 to IX-1981, tree trunks, P. Perkins and D. Young. 1 ♂, 2 ♀ (Liberty Co.), Torreya State Park, 19-V-1971, light trap, G. Fairchild and H. Weems. 1 ♀ (Marion Co.), 7 km S of Ft. McCoy, 7-VIII-1971, tree trunk, D. Young and S. Telford. 15 ♂, 35 ♀ (Wakulla Co.), Wakulla Springs, 1-V-1980, light and flight traps, D. Young. *Georgia*—94 ♂, 16 ♀ (Chatham Co.), Ossabaw Island, 3-4-VIII-1983, light traps, R. Roberts. 1 ♂ (Clinch Co.), Edith, 13-IX-1975, tree trunk, D. Young. *Louisiana*—1 ♂ (DeSoto Parrish), Mansfield, VIII-1971, light trap, J. Reinert. *North Carolina*—19 ♂, 11 ♀ (Hoke Co.), Ft. Bragg, 7-V to IX-1979, light traps, R. Intermill. *South Carolina*—50 ♂, 28 ♀ (Richmond Co.), Ft. Jackson, 3-V to IX-1978, 1979, light traps, R. Intermill.

ADDITIONAL RECORDS (all from Dr. V. Newhouse, in light traps). *Arkansas*—1 ♀ (Union Co.), El Dorado, 21-V-1966. *Florida*—3 ♀ (Collier Co.), Corkscrew Swamp, 11-12-IX-1965. 1 ♀ same data but 9-VI-1967. 11 ♂, 22 ♀ (Dade Co.), Mahogany Swamp, Everglades National Park, all months except for March and May, 1964–1968. 1 ♂, 3 ♀ (Dade Co.), Royal Palm Hammock, Everglades National Park, 15-16-XI-1964; 28-29-IV-1965 and 4-II-1969. *Georgia*—2 ♀ (Ware Co.), Waycross, Laura Walker State Park, 31-VIII-1965. 1 ♂, 1 ♀ (Charlton Co.), Billy's Island, Okefenokee Swamp, 8-X-1965. *Louisiana*—1 ♀ (Orleans Parrish), Kenner, 4-VIII-1966. 1 ♀ (Orleans Parrish), New Orleans, 25-X-1966. *Maryland*—4 ♂, 1 ♀ (Anne Arundel Co.), Annapolis,22-IX-1966. 1 ♀ (Wicomico Co.), Willards, 4-VIII-1968. *Mississippi*—2 ♂, 37 ♀ (Hancock Co.), Bay St. Louis, 26-IV; 30-VIII; 4–28 to IX-19 and 5-X-1967. *North Carolina*—1 ♂, 1 ♀ (Jones Co.), Trenton, 17-IX-1965. 1 ♂, 1 ♀ (Onslow Co.), Jacksonville, 9-19-IX-1965. 1 ♀ (Duplin Co.), Rose Hill, 4-5-VIII-1965.

DISCUSSION: Only selected references to *L. shannoni* are given here. Forattini (1973), Martins et al. (1978) and Young (1979) provide additional references and synonyms.

There are no records of this species in Texas or in northern Mexico, north of Puebla State (Martins et al. 1978). The magnitude of this distributional gap may be partly due to the lack of collecting but, more likely, it reflects the absence of extensive hardwood forests, the pre-

ferred macrohabitat of this species. Future collections in east Texas may reveal its presence there. The new records from Maryland and Delaware represent the known northern distributional limits of *L. shannoni*.

Both sexes of *L. shannoni* from the USA and the neotropics (from southern Mexico to northern Argentina) are remarkably similar in structure. There is little or no morphological variation (Rozeboom 1944). In terms of anthropophilic behavior, however, Fairchild (1955) and others have noted that females bite humans more commonly in the USA and Mexico than elsewhere.

Rosabal and Miller (1970) collected 530 males and 233 females from tree hollows in Louisiana. No parasites were observed in 29 females dissected. Perkins (1982) dissected 414 male and 166 female *L. shannoni* from 2 localities in Florida (Gulf and San Felasco Hammocks) and found acephaline gregarines in 3 males (0.7%) and 15 females (9.1%). Three mites, provisionally identified as *Eustigmaeus* sp., were found attached to the abdomens of 2 female flies. The ovary of one female was parasitized by a small unidentified nematode. One female *L. shannoni* collected on 17-VII-1981, Gulf Hammock, had thousands of relatively large unidentified flagellates in the midgut and foregut but not in the mouthparts. The fly was dissected 9 days after it had fed on man, but no signs of human disease were subsequently observed. The midgut of a dissected male *L. shannoni* from the same locality contained relatively small flagellates that were probably monoxenous. Further studies are being made to characterize these flagellate infections.

Perkins (1982) established a laboratory colony of *L. shannoni* from field-collected females in Florida (Endris et al. 1982) and provided information on its life cycle and biology.

Females normally deposit as many as 40 viable eggs without having had a previous bloodmeal. Only 12 (3.4%) of 349 reared, nonbloodfed females failed to develop fully formed eggs by the fourth day following emergence. Autogenous females with developing eggs also took multiple bloodmeals between ovipositions, beginning 24 hours post-emergence. Laboratory hosts included man, pig, hamster, squirrel (*Sciurus*), calf, horse, opossum (*Didelphis*) and dog (Endris et al. 1982). Christensen and deVasquez (1982), using the precipitin test for bloodmeal determination in Panama, demonstrated that *L. shannoni* feeds on a variety of mammals, some birds but no reptiles. Females fed on sloths more than any other mammal species, and the authors suggested that *L. shannoni* may play a role in the transmission of *Leishmania braziliensis* among these reservoir hosts in Panama.

Lutzomyia shannoni has not been found in the endemic focus of *Leishmania mexicana* in Texas, but experimentally-infected *L. shannoni* females (WRAIR strain 411 from Texas) transmitted the parasite to non-infected hamsters by bite (Lawyer and Young, unpublished data). Perkins (1982) reported that 26/27 lab-reared females became infected with this strain after feeding on infected hamsters. Zeledón and Alfaro (1973) found unidentified promastigotes in 4/117 (3.4%) wild-caught *L. shannoni* females in Costa Rica, and later Zeledón et al. (1979) identified other flagellates recovered from this sand fly as *Endotrypanum* sp. and *Leishmania herreri*, both sloth parasites.

7. *Lutzomyia (Psathyromyia) tanyopsis* Young and Perkins, n.sp., Fig. 8.

Holotype ♀ (measurements in mm). Wing length 2.28; width 0.66. Most of insect lightly pigmented, pleuron slightly paler than scutum. Head from vertex to distal end of clypeus 0.50 high; 0.38 wide. Eyes small, separated by 0.15 or by a distance = to about 6.8 facet diameters. Flagellomere I 0.27 long, combined length of II + III = 0.23; ascoids present on all flagellomeres except last (flag. XIV), each ascoid with a long proximal spur. Labrum 0.34 long. Lengths of palpomeres: 1, 0.05; 2, 0.17; 3, 0.19; 4, 0.13; 5, 0.31; with about 12 sensilla at middle of palpomere 3. Cibarium with 4 short equidistantly-spaced horizontal teeth, middle pair inwardly directed and outer pair slanted outwardly; an irregular row of about 10 small vertical teeth present; cibarial arch complete but more diffuse in middle; pigment patch subtriangular, wider posteriorly. Pharynx 0.20 long, without spines. Pleuron with 6 upper and 4 lower episternal setae. Lengths of wing vein sections: *alpha** 0.36; *beta* 0.34; *delta* 0.04; *gamma* 0.47. Lengths of femora, tibiae and basitarsi as follows: foreleg, 0.85, 0.83, 0.50; midleg, 0.88, 1.02, 0.54; hindleg, 0.98, 1.32, 0.63. Spermathecae spherical with basal annulations; individual ducts smooth walled, about 8X length of common duct.

KNOWN DISTRIBUTION: USA. *Arizona* (Pima Co.), Fig. 20.

MATERIAL EXAMINED: USA. *Arizona—Holo-*

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* *alpha* = Length of R_2 from its junction with R_3 to costa (see Fig 2B).

beta = Length of R from junction of R_2 and R_3 to junction with R_4

delta = Length of R_1 extending beyond junction of R_2 and R_3.

gamma = Length of R from origin of R_5 to origin of R_{2+3} and R_4.

type ♀ (Pima Co), Sabino Canyon, Coronado National Forest, 9-VIII-1953, light trap, G. Butler (USNM). *Paratypes*, 2 ♀, same data.

DISCUSSION: We tentatively place *L. tanyopsis* in the subgenus *Psathyromyia* because of the long proximal spurs of the antennal ascoids and the cibarial armature. The spermathecae, however, differ in structure from those of other *Psathyromyia* females and from those of the neotropical species *L. aclydifera* (Fairchild and Hertig), *L. dreisbachi* (Causey and Damasceno) and *L. hermanlenti* (Martins, daSilva and Falcão), that also have long proximal spurs on the ascoids. The undiscovered male of *L. tanyopsis* also probably will have ascoidal spurs and other features that should readily associate it with the female and determine its definite placement in the genus.

The specific name, "tanyopsis," is a Greek word meaning long face or long in appearance (see Fig. 8A).

SPECIES GROUP *ARAGAOI* THEODOR

8. *Lutzomyia texana* (Dampf), Figs. 9, 16 and 17.

Phlebotomus texanus Dampf 1938:119 (♂, ♀, San Antonio, Bexar Co., Texas). Rozeboom 1944:274 (listed). Addis 1945a:328 (keyed). Packchanian 1946:37 (listed). Barretto 1947:226 (refs.). Thurman et al. 1949:199 (listed). Vargas and Díaz Nájera 1953b:313 (Mexican records). Fairchild and Hertig 1953:21 (keyed). Fairchild 1955:195 (listed). Quate 1955:242 (keyed, refs.); 1965:92 (listed). Eads et al. 1965:251 (coll. data, Texas). Easton et al. 1968:466 (Kinney Co., Texas).

Lutzomyia texana: Barretto 1962:99 (listed). Theodor 1965:186 (classif.). Rosabal and Miller 1970:180 (keyed). Young 1972:61 (coll. data, Texas). Lewis 1975:502 (mouthpart morphol.). Calisher et al. 1977:1000 (mention). Eads 1978:540 (refs.). Chaniotis 1978:19 (mention). Martins et al. 1978:146 (dist.). Young 1979:133 (compared to *L. barrettoi*).

Psychodopygus texanus: Forattini 1971:105 (listed); 1973:480 (tax.).

KNOWN DISTRIBUTION: Honduras, Mexico, USA. *Texas* (Aransas, Bexar, Cameron, Edwards, Gillespie, Kinney, San Patricio, Uvalde and Val Verde counties), Fig. 20.

MATERIAL EXAMINED: USA. *Texas*—1 ♂, 9 ♀ (Aransas Co.), Aransas Wildlife Refuge, 22-IV-1966, W. Wirth and R. Jones. 2 ♂, 2 ♀ (Cameron Co.) near Brownsville, 28-I-1963 and II-1963, 1964, light traps, R. Eads. 1 ♂, 3 ♀ (Edwards Co.), 6 km N of Barksdale, VI-1982, armadillo burrow, R. Endris. 2 ♂, 2 ♀ (Cameron Co.), near Brownsville, 28-I-1963 and II-1963, 1964, light traps, R. Eads. 15 ♂, 24 ♀ (Gillespie Co.), Fredericksburg, V-IX-1968, light traps, F. Blanton and H. Borchers. 32 ♂, 45 ♀ (San Patricio Co.), Welder Wildlife Refuge, 5-XII-1965, armadillo burrow, D. Young. 4 ♂, 13 ♀, same data but 29-V-1980, D. Young et al. 6 ♂, 9 ♀ (Uvalde Co.), Garner State Park, 18-V-1965, light trap, D. Young. 6 ♂, 12 ♀, same data but VI-1982, light trap, armadillo burrow and in latrine. 5 ♂, 4 ♀ (Val Verde Co.), Del Rio, IX-X-1963, light traps, R. Eads. Mexico. *Morelos*—2 ♂, 71 ♀, Miacatlán, 9-IV to 5-VI-1980, flight trap, M. Camino. *San Luis Potosi*—El Salto Falls, 17-VI-1969, light trap, W. Hasse. Honduras. *Copan*—1 ♂, 1 ♀, Santa Rosa, IV-1964 and 5-V-1966, light traps, F. Blanton and J. Matta. *Comayagua*—1 ♂, Siguatepeque, IV-1967, light trap, F. Blanton.

DISCUSSION. *Lutzomyia texana* is one of the largest species in North America. The wing length of females ranges from 2.55 to 2.93 mm (n = 9) and of the males from 2.30 to 2.57 mm (n = 9) (all from Texas).

The poorly preserved specimens from Honduras were tentatively identified as *L. texana*. They might actually represent *L. barrettoi majuscula* Young, a closely related species occurring in Colombia and Central America. It has not been established where, or if populations of these 2 species meet in Central America. Structural differences between these taxa are slight and may reflect intraspecific variation of a single widespread species.

Lutzomyia texana was first collected in a nest of the leaf-cutting ant, *Atta texana*, near San Antonio, Texas (Dampf 1938). Eads et al. (1965) excavated a number of *Atta* nests near Brownsville, Texas, but failed to recover larvae or resting adults of *L. texana*. Light trap catches by these authors, however, showed that adults were present throughout the year near Brownsville, but not in the Del Rio, Texas area.

Young (1972) reported that armadillo burrows serve as the usual resting sites for this species in Texas. In June 1980, one of us found 9 recently bloodfed females in a burrow inhabited by armadillos near Sinton, Texas, but there is no direct evidence that the flies had fed on these mammals. The flies died before their eggs were laid.

SUBGENUS *HELCOCYRTOMYIA* BARRETTO

9. *Lutzomyia (Helcocyrtomyia) apache* (Young and Perkins n.sp., Figs. 10, 16 and 17.

Holotype ♂ (measurements in mm). Wing length 2.47; width 0.66. Head, scutum, abdominal sclerites and external genitalia infuscated; rest of body pale or nearly pale. Head from vertex to tip of clypeus, 0.41 high; 0.39 wide. Eyes separated by 0.15 or by a distance = to about 8 facet diameters. Flagellomere 1, 0.27

long, combined length of II + III, 0.25; ascoids simple, present on all flagellomeres except last (XIV); ascoids on II reaching to or beyond end of flagellomere. Labrum 0.19 long. Lengths of palpomeres: 1, 0.04; 2, 0.12; 3, 0.18; 4, 0.15; 5, 0.34; about 8 palpal sensilla scattered on middle half of palpomere 3. Cibarium without visible teeth; pigment patch narrow and faint; cibarial arch diffuse, more conspicuous at sides. Pharynx 0.16 long, with posterior ridges but no spines. Pleuron with 5–6 upper and 4–5 lower episternal setae. Lengths of wing vein sections: *alpha* 0.46; *beta* 0.35; *delta* 0.13; *gamma* 0.50. Lengths of femora, tibiae and basitarsi as follows: foreleg, 0.89, 0.86, 0.51; midleg, 0.87, 1.00, 0.58; hindleg, 0.95, 1.34, basitarsus missing; hind femur without spines. Terminalia. Gonostyle 0.24 long with 5 spines, basal pair inserted at different levels, isolated median spine closer to basal pair than to terminal pair, no subterminal seta. Gonocoxite 0.31 long, with basal tuft of 5 slender setae and about 12 scattered distal setae. Paramere simple, with dorsal setae on at least half its length. Aedeagus slender, well-pigmented with acute tip. Ejaculatory apodeme and sperm pump 0.25 long, each aedeagal filament 1.20 long or about 4.8X length of ejaculatory apodeme and sperm pump, the distal end inflated subterminally as shown in Fig. 10B. Lateral lobe 0.47 long.

Allotype ♀ (measurements in mm). Wing length 2.18; width 0.56. Coloration as in ♀. Head 0.39 high; 0.33 wide. Eyes separated by 0.15 or by a distance = to about 8.5 facet diameters. Flagellomere I, 0.23 long, combined length of II + III, 0.22; ascoids simple, on all flagellomeres except last (XIV), tips of those on flag. II extending beyond end of flagellomere. Labrum 0.19 long. Lengths of palpomeres: 1, 0.04; 2, 0.11; 3, 0.15; 4, 0.13; 5, 0.31; palpal sensilla as in m. Cibarium with 4 sharp, equidistantly-spaced horizontal teeth and a row of 10–14 small vertical teeth; cibarial arch conspicuous at sides, nearly invisible in middle; pigment patch subtriangular, faint. Pharynx 0.16 long, without spines. Pleuron with 4 upper and 2–3 lower episternal setae. Lengths of wing vein sections: *alpha* 0.39; *beta* 0.34; *delta* 0.05; *gamma* 0.37. Lengths of femora, tibiae and basitarsi: foreleg, 0.73, 0.71, 0.44; midleg, 0.76, 0.93, 0.51; hindleg, 0.86, 1.13, 0.69; femora without spines. Spermathecae button-like as shown in Fig. 10F with long, slender individual ducts, each about 25X length of common duct or greater than 5X length of stem of genital fork.

KNOWN DISTRIBUTION: USA. *Arizona* (Apache, Cochise and Gila counties), Fig. 20.

MATERIAL EXAMINED: USA. *Arizona*—holotype ♂ (Apache Co.), Springerville, 3-VII-1953, light trap, W. Wirth. ♀ *allotype* (Cochise Co.), 3.1 km SW of Portal, V-VI-1967, blacklight trap, C. Sabrosky. Other *paratypes*, 12 ♂, 11 ♀, same data as holotype. 2 ♀, same data as allotype. 2 ♀ (Gila Co.), 27 km NE of Payson, Tonto Creek, 10-VIII-1978, light trap, C. Ray (UCR).

DISCUSSION: Structural similarities indicate that *L. apache* is closely related to *L. vexator* and *L. oppidana*, both of which may occur with it in the USA and northern Mexico. Both sexes of *L. apache* can be separated from those of the other species by the characters given in the keys. *Lutzomyia vindicator* (Dampf), known from Mexico, differs from *L. apache* by its broader, differently shaped paramere, shorter aedeagal filaments (less than 4X the length of the ejaculatory apodeme and sperm pump) and by the wider shorter individual sperm ducts of the female. The cibarial armatures of these females are remarkably similar.

The wing length of 8 female paratypes of *L. apache* ranges from 2.42 to 2.88 mm; that of 5 male paratypes ranges from 2.37 to 2.55 mm.

The specific name, "apache," refers to the tribe of American indians living in the southwestern USA and northern Mexico.

10. *Lutzomyia (Helcocyrtomyia) oppidana* (Dampf), Figs. 11, 16 and 17.

Phlebotomus oppidanus Dampf 1944:247 (♀, San Jacinto, Federal District, Mexico). Barretto 1947:215 (listed). Vargas and Díaz Nájera 1953b:312 (listed). Fairchild and Hertig 1957:325 (♀, Nuevo León State, Mexico, keyed); 1959:124 (listed). Fairchild and Harwood 1961:240 (coll. data, Adams Co., Washington, keyed). Fairchild and Hertig 1961:26 (compared to *L. vargasi* ♂). Ortiz and Alvarez 1963:312 (listed). Harwood 1965:1 (coll. records, Washington State and British Columbia, rearing). Eads et al. 1965:251 (listed). Ortiz 1965b:25 (listed). Quate 1965:92 (listed). Easton et al. 1967:429 (Presidio Co., Texas). Easton et al. 1968:467 (Presidio Co., Texas).

Lutzomyia oppidana: Barretto 1962:96 (listed). Theodor 1965:183 (listed). Rosabal and Miller 1970:780 (keyed). Forattini 1971:99 (listed). Young 1972:61 (listed). Downes 1972:1135 (listed). Martins and Morales-Farias 1972:365 (Listed). Forattini 1973:209 (redescript., keyed). Chaniotis 1974:334 (Ravalli Co., Montana); 1978:19 (mention). Eads 1978:541 (Larimer Co., Colorado). Martins et al. 1978:77 (dist.).

KNOWN DISTRIBUTION: Mexico. USA. *Colorado* (Fort Collins and Larimer counties), *Montana* (Ravalli Co.), *Texas* (Presidio Co.) and *Washington* (Adams and Whitman counties). Canada. *Alberta* and *British Columbia*, Fig. 21.

MATERIAL EXAMINED: USA. *Colorado*—16 ♂, 18 ♀ (Fort Collins Co.), approx. 400 m east of Horsetooth Reservoir, 18-VIII- and 8-IX-1983, oil and light traps, C. Moore and B. Francy. *Montana*—1 ♀ (Ravalli Co.), 8 km E of Hamilton., Rock pile, 22-VIII-1973, B. Chaniotis. *Washington*—6 ♂, 15 ♀ (Adams Co.), Othello, Columbia Wildlife Refuge, 25-V to 9-VIII-1960, R. Harwood. Mexico. 30 ♂, 9 ♀ (*Nuevo León*), 32 km NW of Monterrey, 9-IX-1955, rock crevices, P. Galindo and H. Trapido.

DISCUSSION: The habits of *L. oppidana* are poorly known. Harwood (1965) reported that females in the laboratory fed on snakes and lizards, but not on frogs, white mice or humans. Diurnal resting sites include animal burrows, small caves and rock crevices.

This species apparently has a widespread geographic distribution in the western USA and Mexico.

11. *Lutzomyia (Helcocyrtomyia) stewarti* (Mangabeira and Galindo), Figs. 12, 16 and 17.

Phlebotomus stewarti Mangabeira and Galindo 1944:185 (♂, ♀, Livermore, Alameda Co., California). Rozeboom 1944:274 (listed). Addis 1945a:328 (keyed). Packchanian 1946:37 (dist.). Barretto 1947:225 (catalogued). Thurman et al. 1949:199 (listed). Vargas and Díaz Nájera 1953b:312 (Sonora State, Mexico). Quate 1955:243 (refs.). Fairchild 1955:194 (listed). Fairchild and Hertig 1957:326 (keyed, refs.). Lauret 1958:319 (coll. data, California). Ortiz and Alvarez 1963:312 (listed). Eads et al. 1965:251 (listed). Ortiz 1965b:25 (listed). Quate 1965:92 (listed). Chaniotis 1967:221 (rearing data, lab. hosts, biology). Chaniotis and Anderson 1968:273 (coll. data, pop. dynamics, California).

Lutzomyia stewarti: Barretto 1962:96 (listed). Theodor 1965:183 (listed). Rosabal and Miller 1970:180 (keyed). Ayala and Lee 1970:891 (as host for saurian malaria). Ayala 1971c:598 (as host for saurian malaria); 1973:266 (disease relationships, California). Forattini 1973:270 (tax.). Ayala 1977:275 (mention). Martins et al. 1978:81 (dist.). Eads 1978:540 (listed, refs.). Young et al. 1983:315 (Riverside Co., California).

KNOWN DISTRIBUTION: Mexico. USA. *California* (Alameda, Contra Costa, Kern, Mendocino, Monterey, Riverside, San Diego, San Mateo and Yolo counties). Fig. 21.

MATERIAL EXAMINED: USA. *California*—1 ♂ paratype (Contra Costa Co.), Marsh Creek, 21-IX-1943, P. Galindo and O. Mangabeira. 1 ♀ (Mendocino Co.) Ukiah, 29-VII-1948, light trap, W. Wirth. 2 ♂ (Monterey Co.), Ft. Hunter Liggett, 26-V-1981 and 24-VII-1981, light trap. 2 ♂, 5 ♀ (Riverside Co.), 5.6 km S of Palm Desert, 11-13-VIII-1969 and 31-I to 23-II-1970, Malaise trap, S. Frommer. 1 ♂, 3 ♀ (Riverside Co.), Deep Canyon, Coyote Creek, 10-17-V-1975, Malaise trap, S. Frommer and R. Worley, 1 ♂ (Riverside Co.), Rancho Mirage, Magnesia Springs Canyon, 16-VI-1981, CO_2 trap, R. Brenner.

DISCUSSION: A female of *L. stewarti* was identified from Ukiah, Mendocino Co., California. This specimen was provisionally, but incorrectly, determined earlier as *L. californica* by Fairchild and Hertig (1957) and was cited as that species by Chaniotis and Anderson (1968).

Lauret (1965) collected 154 adults of *L. stewarti* in light traps from 1 June to 10 October 1956 at Woodside, San Mateo Co., California. Chaniotis (1967) stated that in California adults of *L. stewarti* are active for about 6 months of the year and that the fourth instar larvae diapause during the winter months.

Adults rest, and perhaps breed, in the burrows of ground squirrels, *Citellus* spp., where they feed on lizards and snakes (Chaniotis and Anderson 1968).

Chaniotis (1967) reared *L. stewarti* in the laboratory and provided data on its life cycle, feeding and mating behavior, and other habits. Ayala and Lee (1970) observed sporozoites of *Plasmodium mexicanum*, a saurian malaria, in wild-caught *L. stewarti* after they were experimentally infected in the laboratory.

12. *Lutzomyia (Helcocyrtomyia) vexator* (Coquillett), Figs. 8 and 13.

Phlebotomus vexator Coquillett 1907:102 (♂, ♀, Plummer's Island, Montgomery Co., Maryland). Shannon 1913:165 (biology, Virginia record). Shannon 1926:193 (♂, ♀). Dyar 1929:113 (tax.). Theodor 1933:274 (♂, ♀, tax.). Hall 1936:29 (Jackson Parish, Louisiana). Dampf 1938:122 (mention). Mangabeira and Galindo 1944:183 (♂, ♀, redescript., California records). Rozeboom 1944:274 (listed). Dampf 1944:237 (Sonora State, Mexico). Addis 1945:328 (keyed). Packchanian 1946:37 (dist., Virginia record). Dampf 1947:205 (tax.). Barretto 1947:224 (full refs.). Thurman 1949:199 (listed). Vargas and Díaz Nájera 1953b:313 (Sonora State, Mexico). Quate 1955:243 (keyed, refs.). Fairchild 1955:191 (classif.). Fairchild and Hertig 1957:327 (keyed, tax., refs.). Hertig and Johnson 1961:753 (mention). Ortiz and Alvarez 1963:312 (listed). Roberts 1965:28) (Mississippi record). Ortiz 1965:25 (listed). Quate 1965:92 (listed). Downes 1972:1135 (Perth, Ontario, Canada).

Phlebotomus vexator occidentis Fairchild and Hertig 1957:334 (♂, ♀, Alturas trap station, Modoc Co., California and Topaz Lake, Mono Co., California). Fairchild and Harwood

1961:240 (keyed, Adams Co., Washington). Ortiz and Alvarez 1963:312 (listed). Chaniotis and Anderson 1964:27 (descript. immatures, rearing data, tax. California). Harwood 1965:1 (ecology, Washington; Alberta, Canada record). Eads et al. 1965:251 (listed). Quate 1965:92 (listed). Chaniotis 1967:221 (rearing data, biology, California). Anderson and Ayala 1968:1023 (as vector of toad trypanosome, California). Chaniotis and Anderson 1968:273 (pop. dynamics, redescript., California records, nat. infected with trypanosomes). Ayala 1970a:387 (infected with hemogregarines) Shemanchuk et al. 1978:1355 (Alberta, Canada in marmot burrows).

Lutzomyia vexator (or as *vexatrix*) *occidentis*: Barretto 1962:96 (listed). Rosabal and Miller 1970:180 (keyed). Ayala and Lee 1970:891 (as host for saurian malaria). Ayala 1970c:13 (development of malaria in females); 1971a:440 (infected with gregarines); 1971b:433 (saurian malaria infected with trypanosomes); 1971c:598 (saurian malaria). Young 1972:62 (Texas records). Ayala 1973:266 (parasite relationships). Chaniotis 1974:334 (Montana record). Eads 1978:540 (refs.). Martins et al. 1978:77 (dist.). Chaniotis 1978:19 (mention).

Lutzomyia vexator (or as *vexatrix*): Barretto 1962:96 (listed). Theodor 1965:183 (classif.). Quate 1965:92 (listed). Rosabal and Miller 1970:180 (keyed, Louisiana records). Young 1972:63 (Florida records). Forattini 1973:272 (tax.). Lewis 1975:501 (mouthpart morphol.). Aitken et al. 1977:582 (Connecticut records). Martins et al. 1978:77 (dist., including Mann Co., Calif). Chaniotis 1978:19 (mention). Eads 1978:540 (refs.). Steyskal 1979:423 ("*vexator*" as correct spelling). Young et al. 1981:446 (mention). Endris et al. 1982:401 (rearing data).

Known distribution: Mexico. Canada. *Alberta* and *Ontario*. USA. *Alabama* (Lauderdale Co.). *Arkansas* (Garland Co.). *California* (Alameda, Contra Costa, Kern, Mann, Marin, Mendocino, Modoc, Mono, Monterey, Riverside, San Luis Obispo, San Mateo, Solano, Ventura and Yolo counties). *Colorado* (El Paso Co.). *Connecticut* (Middlesex Co.). *Florida* (Alachua, Collier, Highlands, Levy and Wakulla counties). *Georgia* (Gwinnett Co.). *Louisiana* (Jackson, Morehouse and Orleans parishes). *Maryland* (Montgomery Co.). *Mississippi* (Washington Co.). *Montana* (Ravalli Co.). *New Mexico* (Eddy and Rio Arriba counties). *Oklahoma* (Caddo and Oklahoma counties). *Texas* (Edwards, Gillespie, Presidio and Uvalde counties). *Virginia* (Augusta and Fauquier counties). *Washington* (Adams and Whitman counties). *Wyoming* (Goshen Co.). Fig. 22.

Material examined: USA. *Arkansas*—3 ♂, 2 ♀ (Garland Co.), Hot Springs, VIII-1971, light trap, J. Reinert. *California*—1 ♀ (Alameda Co.), Strawberry Canyon, X-1948, light trap, W. Wirth. 1 ♂, 1 ♀ (Contra Costa Co.), Marsh creek, 15-19-IX-1943, P. Galindo et al. 1 ♀ (Modoc Co.), Alturas Trap Station, VIII-1948, light trap, R. Coleman. 1 ♀ (Mono Co.), Topaz Lake, 1-VIII-1948, light trap, R. Coleman. 8 ♂, 20 ♀ (Monterey Co.), Ft. Hunter Ligget, 19-V to 25-IX-1981, light trap, U.S. Army personnel. 1 ♂ (Riverside Co), Riverside, 20-21-VII-1071, light trap, S. Frommer and L. Luna. *Colorado*—1 ♀ (El Paso Co.), Ft. Carson, VIII-1981, light trap, U.S. Army personnel. *Connecticut*—1 ♀ (Middlesex Co.), North Madison, 8-VII-1977, light trap, T. Aitken and K. Kloter. 1 ♂ (Middlesex Co.), Millington, VII-1977, light trap, T. Aitken. *Florida*—4 ♂, 2 ♀ (Alachua Co.), San Felasco Hammock, various dates in V, VIII and IX-1967, 1968, light and flight traps, F. Blanton, D. Young and G. Fairchild. 2 ♂, 2 ♀ (Levy Co.), Gulf Hammock, IX-1975, under loose bark of standing dead trees, D. Young. 22 ♂, 30 ♀, same data but 8-V to 15-IX-1981, P. Perkins and D. Young. 1 ♀ (Wakulla Co.), Wakulla Springs, 1-V-1980, light trap, W. Kramer. *Maryland*—1 ♂, 1 ♀ (Montgomery Co.), Plummer's Island, VII, R. Shannon. 2 ♂, 4 ♀ but reared, 1932–1936, M. Hertig. 1 ♂, 4 ♀, same data but 6-7-VIII-1949, hollow tree and in house, M. Hertig. *Oklahoma*—1 ♂, 1 ♀ (Caddo Co.), Anadarko, VIII-1971, light trap, J. Reinert. 1 ♂ (drawing of style, specimen not seen), Edmond (Oklahoma Co.), no other data. *Texas*—2 ♂, 1 ♀ (Edwards Co.), 6 km N of Barksdale, V-1982, at light, P. Lawyer et al. 2 ♂, 7 ♀ (Gillespie Co.), Fredericksburg, V, VII and IX-1968, 1969, light traps, H. Borchers. 2 ♂, 3 ♀ (Uvalde Co.), Garner State Park, 18-V-1965, light traps, D. Young. *Washington*—27 ♂, 36 ♀ (Adams Co.), Othello, Columbia Wildlife Refuge, 25-VI to 30-X-1960, marmot burrows, R. Harwood. *Wyoming*—1 ♀ (Goshen Co.), Lingle, VIII-1970, light trap, M. Butler.

Additional records (all collected by V. Newhouse in light traps): *Florida*—1 ♂ (Collier Co.), Turner River Jungle Garden, 30-VII-1963. 6 ♀ (Collier Co.), Corkscrew Swamp, 22-23-I-1965. 5 ♂, 3 ♀, same data but 9-VI-1967. 1 ♂, 11 ♀, same data but 3-4-VIII-1967. 2 ♂, 4 ♀, same data but 1-12-IX-1967. 1 ♂, 2 ♀ (Highlands Co.), Brighton Indian Reservation, 13-18-III-1966. *Louisiana*—1 ♀ (Orleans Parish), Kenner, 4-VIII-1966. *New Mexico*—1 ♀ (Eddy Co.), Carlsbad, Rattlesnake Springs, 17-VII-1968. 2 ♀ (Rio Arriba Co.), Albiquin, 19-20-VIII-1965

Discussion: Fairchild and Hertig (1957) studied males and females of *L. vexator* from California and concluded that they differed

subspecifically from specimens collected on Plummer's Island, Maryland, the type-locality. They stated that the California specimens, which they named *L. vexator occidentis*, had the following distinguishing characteristics: "Male with genital filaments shorter, less than 4 times length of pump; style shorter and stouter, the two subapical spines farther apart and the most basal one of this pair closer to the basal spine than in *vexator*. Basal tuft of coxite of 4 setae. Female with spermathecae shorter, more slender, less than twice as long as stem of genital fork. Cibarium narrower, with but 4 horizontal teeth, lacking the sublateral shorter teeth found in *vexator*."

Chaniotis and Anderson (1964, 1967) redescribed *L. v. occidentis* from California specimens, noting that the aedeagal filaments of 10 males were 4 times the length of the ejaculatory apodeme and sperm pump and that the basal gonocoxal tuft consisted of 4 or 5 long hair-like setae. The length of the aedeagal filaments of Florida males that were examined in this study varied from 3.6 times to 4.6 times the length of the ejaculatory apodeme and sperm pump and the gonocoxal tuft consisted of 4 or 5 setae. This range of measurements encompasses that observed for all other males examined (including California and Washington State specimens) except that one male from Plummer's Island had aedeagal filaments 4.9 times the length of the ejaculatory apodeme and sperm pump. The length of the gonostylus and presence or absence of sublateral teeth in the female cibarium are not diagnostic. Of 7 females examined from the same population in Texas, 2 had sublateral teeth while the other 5 lacked them. The length of the gonostylus of 16 males from Washington State ranged from 0.21 to 0.24 mm and from 0.24 to 0.26 for 3 males from Maryland. In view of this variation and of our inability to detect clear-cut, consistent differences among specimens from various localities, *L. vexator* is treated as a monotypic species that has a widespread geographic distribution in North America.

Lutzomyia vexator is easily reared in the laboratory. Marshall Hertig maintained a closed colony from 1932 to 1936 at Harvard University from specimens collected at Plummer's Island, Maryland. Chaniotis and Anderson (1964) and Chaniotis (1968) successfully reared *L. vexator* and provided information on its biology under laboratory conditions and in the field (Chaniotis and Anderson 1968). A laboratory colony is presently being maintained at the University of Florida (Endris et al. 1982).

Ayala and colleagues (1968–73) published several articles on sand flies (mostly *L. vexator*) and associated parasites in California. There, the sand flies rest in ground squirrel burrows that are also occupied by lizards, snakes and other cold-blooded vertebrates which serve as hosts for these insects.

Sporozoites of *Plasmodium mexicanum* a saurian malaria, were observed in experimentally-infected sand flies (mostly *L. vexator*) from California. These sporozoites were infective to *Scleoporus* lizards when injected intraperitoneally. Transmission was not demonstrated by sand fly bite and no natural infections were found in the presumed vector.

During the past 3 years in Florida, over 600 laboratory-reared females of *L. vexator* that had previously fed on *Plasmodium floridense*-infected anoles (*Anolis carolinensis*) were dissected. Oocysts were seen in over 50% of the flies that were dissected 4–20 days after the infecting bloodmeals, but no sporozoites were detected.

In addition to these preliminary observations, it is noted that naturally infected *Anolis* have been found in several Florida localities where phlebotomine sand flies do not occur. Although hardly conclusive, these observations suggest that *P. floridense* is not transmitted to *Anolis* and fence lizards (*Sceloporus*) in Florida by phlebotomines.

SUBGENUS *MICROPYGOMYIA* BARRETTO

13. *Lutzomyia (Micropygomyia) californica* (Fairchild and Hertig), Fig. 14.

Phlebotomus californicus Fairchild and Hertig 1957:328 (♂, ♀, Ft. Yuma, Imperial Co., California). Fairchild and Harwood 1961:242 (Othello, Adams Co., Washington). Ortiz and Alvarez 1963:312 (listed). Ortiz 1965a:205 (listed). Eads et al. 1965:251 (listed). Quate 1965:92 (listed). Easton et al. 1967:429 (Presidio Co., Texas); 1968:467 (listed). Chaniotis 1967:221 (life cycle, rearing data). Chaniotis and Anderson 1968:273 (pop. dynamics, California records, redescript.).

Lutzomyia californica: Barretto 1962:95 (listed). Theodor 1965:187 (classif.). Rosabal and Miller 1970:180 (keyed). Forattini 1971:101 (listed). Young 1972:61 (mention). Martins and Morales-Farias 1972:365 (dist.). Ayala 1973:268 (mention). Forattini 1973:12 (tax.). Martins et al. 1978:67 (dist.). Chaniotis 1978:19 (mention). Eads 1978:539 (listed, refs.). Young et al. 1983:315 (Riverside Co., California).

KNOWN DISTRIBUTION: USA. *Arizona*—(Pinal Co.). *California* (Imperial, Inyo, Kern, Lassen, Monterey, Riverside, San Diego and Yolo counties). *Texas* (Presidio and Val Verde counties). *Washington* (Adams Co.), Fig. 23.

Material examined: USA. *Arizona*—5 ♀ (Pinal Co.), Superstitition Mountains, 11-III and 11-X-1942, in cave, R. Flock (USNM). *California*—1 ♂ holotype, 1 ♀ allotype (Imperial Co.), Ft. Yuma, 28-IX-1948, light trap, R. Coleman. 1 ♂ (Riverside Co.), P. L. Boyd Desert Research Center, 15-16-X-1969, Malaise trap, S. Frommer and R. Worley. 1 ♀ (Riverside Co.), Deep Canyon, Coyote Creek, 10-17-V-1975, Malaise trap, S. Frommer. 2 ♀ (Riverside Co.), Cahuilla Hills, Palm Desert, 22-VII-1981, CO_2 trap, R. Brenner. 1 ♂, 1 ♀ same data but 16-IX-1981 and 4-XI-1981. *Texas*—1 ♀ (Val Verde Co.), Del Rio, IX-1963, light trap, R. Eads. *Washington*—7 ♂, 13 ♀ (Adams Co.), Othello, Columbia Wildlife Refuge, VI-VIII-1960, marmot burrows, R. Harwood.

Discussion: Fairchild and Hertig (1957) pointed out that *L. californica and L. chiapanensis* (Dampf) are very similar in structure, differing only in the relative lengths of the female sperm ducts and in details of the male terminalia. Now that additional material is available, there is a strong possibility that these differences reflect geographic variation of a single widespread species that occurs from Panama to northwest USA. We have not examined specimens from Mexico, however, and will therefore continue to recognize these species as distinct.

Apart from the longer common sperm duct of the *L. chiapanensis* female there are no other structural differences between it and *L. californica*. There is considerable variation in the number of horizontal teeth in the female cibarium. Chaniotis and Anderson (1968) counted 16–34 such teeth in 10 or more females from California. The Texas female, examined by us, has 20 teeth; 13 females from Othello, Washington have 16–25 teeth. Females referable to *L. chiapanensis* show similar variation. The female holotype of *L. chiapanensis* from Chiapas State, Mexico, has 25 horizontal teeth (Dampf 1947). Specimens from Panama (n = 5), Costa Rica (n = 1) and El Salvador (n = 1) have 20–24, 20 and 28 teeth respectively.

The males of *L. californica* and *L. chiapanensis* are distinguished by the shape and setation of the gonocoxites. Unlike *L. californica*, *Lutzomyia chiapanensis* has a gonocoxite that is noticeably widened basally and has numerous setae distal to the basal tuft.

In California, females of *L. californica* feed on lizards and snakes, probably within mammal burrows which also serve as resting sites for the flies (Chaniotis and Anderson 1968). These authors collected *L. californica* in Yolo Co., California, in low numbers from June 24 to October 12, 1965, and suggested that there are 3 generations per year in that area.

14. *Lutzomyia (Micropygomyia) cubensis* (Fairchild and Trapido), Fig. 15.

Phlebotomus cubensis Fairchild and Trapido 1950:414 (♂, ♀, near Camaguey, Cuba). Fairchild 1955:194 (listed). Ortiz 1965a:290 (♀ keyed). Avila et al. 1969:3 (mention).

Lutzomyia cubensis: Barretto 1962:95 (listed). Theodor 1965:187 (listed). Young 1972:63 (Big Pine Key, Monroe Co., Florida). Martins and Morales-Farias 1972:369 (listed). Forattini 1973:335 (listed, keyed). Lewis 1975:502 (mouthpart morphol.). Eads 1978:540 (listed). Chaniotis 1978:19 (mention). Martins et al. 1978:62 (dist.).

Known distribution: Cuba. USA. *Florida* (Monroe Co.), Fig. 23.

Material examined: USA. *Florida*—1 ♀ (Monroe Co.), Big Pine Key, 16-I-1969, flight trap, A. Gurney (USNM). 2 ♂, 2 ♀ (Monroe Co.), Middle Torch Key, 13-II-1978, flight trap, W. Wirth. Cuba. 9 ♂, 8 ♀ (holotype, allotype and paratypes), Camaguey and Chapaste, V-VI-1949, tree buttresses, G. Fairchild and H. Trapido.

Discussion: *Lutzomyia cubensis* is a small sand fly that successfully colonized some of the Florida Keys presumably from West Indian stock. Its close relatives, *L. cayennensis* (Floch and Abonnenc) and allies, feed mostly on lizards.

CONCLUDING REMARKS

Much more information on sand flies in North America is needed to understand the extent of their geographic ranges, bionomics and disease relationships. With few exceptions, most observations and collections of these flies were made incidental to other insect studies. The phlebotomine fauna of the western USA is richer than that of the eastern states and it is there that additional species are likely to be discovered and where studies on the known species are especially needed.

ACKNOWLEDGMENTS

We wish to thank Dr. G. B. Fairchild and Dr. Ronald A. Ward for the critical reviews of this manuscript. We also wish to thank Olimpia Arezaga for her excellent work in typing the original and revised manuscripts.

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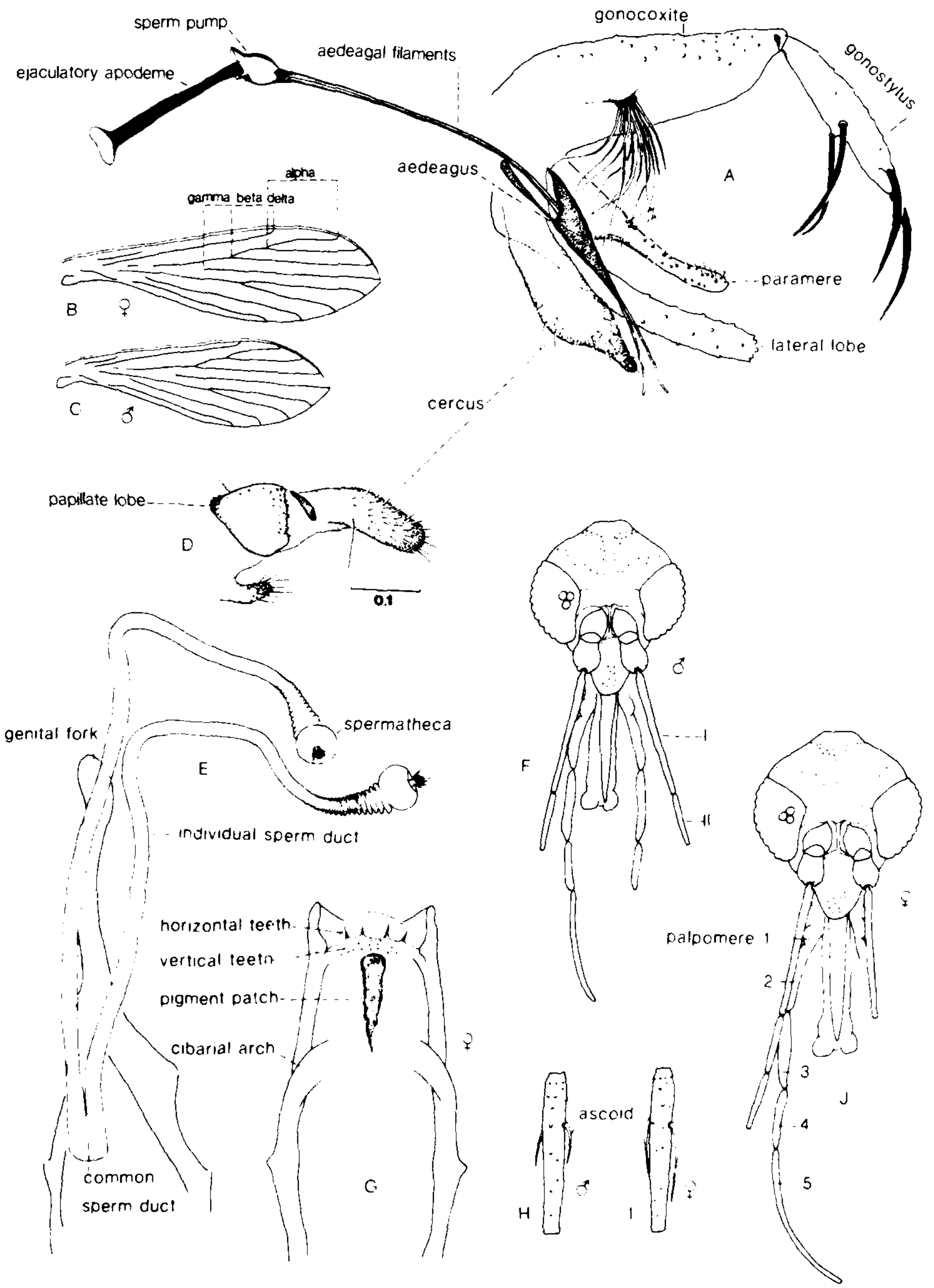

Fig. 2. *Lutzomyia cruciata* (Coquillett). A. Male terminalia, lateral view; B. Female wing; C. Male wing; D. End of female abdomen showing papillate lobe of tergite 8, scale in mm; E. Spermathecae and genital fork; F. Male head; G. Female cibarium; H. Male flagellomere II; I. Female flagellomere II; J. Female head. Drawn from Florida specimens at same scale as comparable structures in Fig. 4.

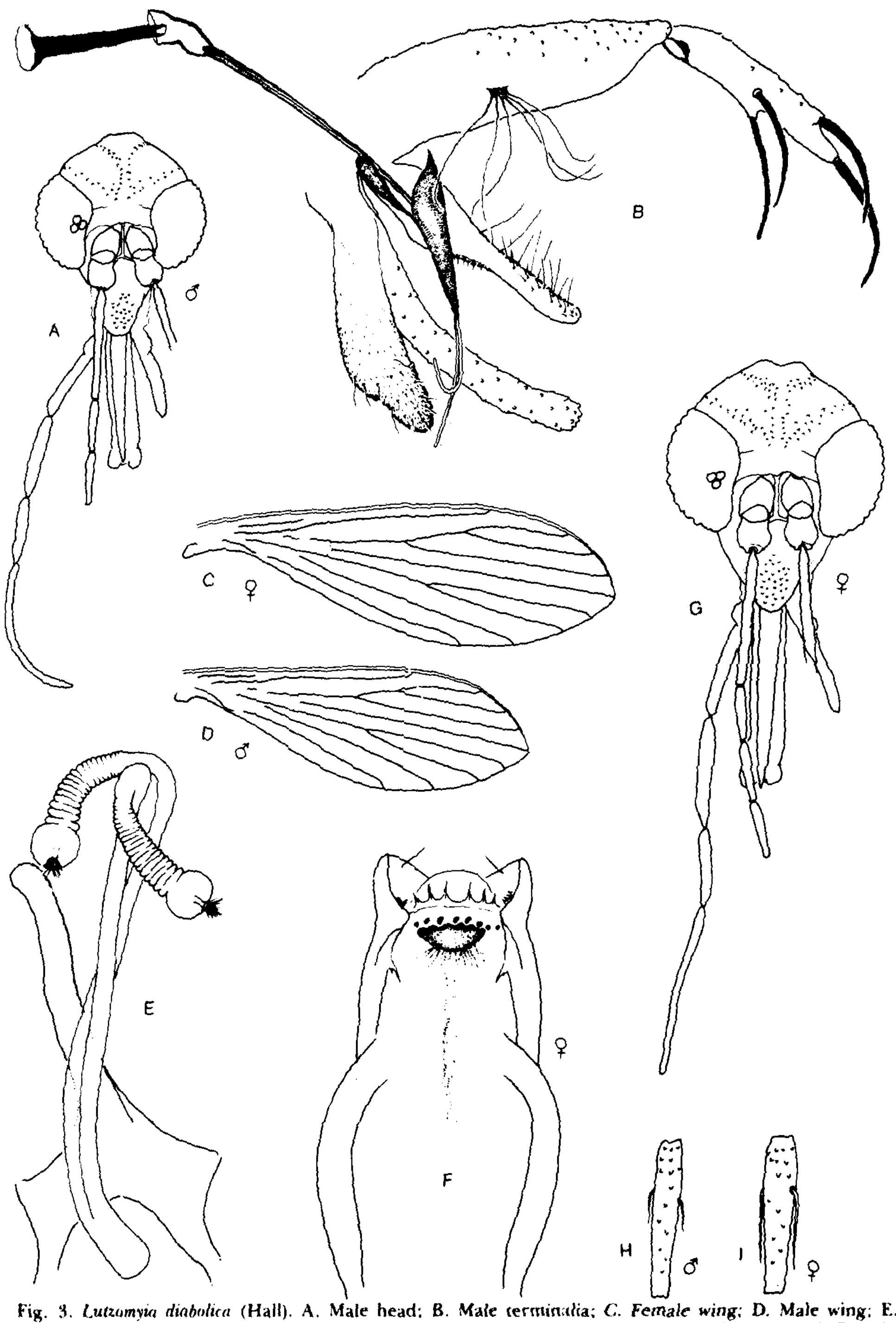

Fig. 3. *Lutzomyia diabolica* (Hall). A. Male head; B. Male terminalia; C. Female wing; D. Male wing; E. Spermathecae and genital fork; F. Female cibarium; G. Female head; H. Male flagellomere II; I. Female flagellomere II. Drawn from Texas specimens at same scale as comparable structures in Fig. 4.

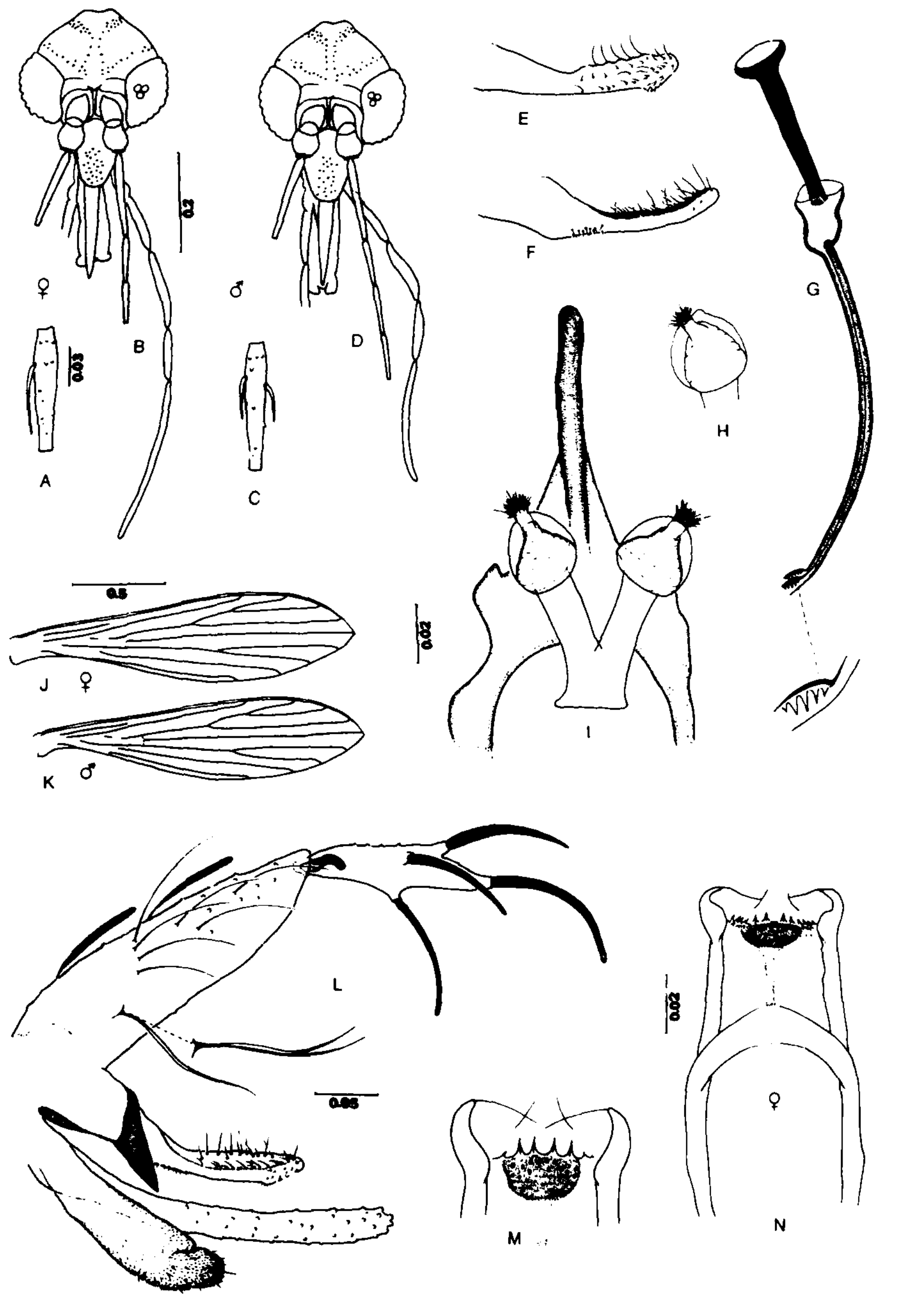

Fig. 4. *Lutzomyia xerophila* Young, Brenner and Wargo. A. Female flagellomere II; B. Female head; C. Male flagellomere II; D. Male head; E. and F. Different views of male parameres; G. Male aedeagal filaments, ejaculatory apodeme and sperm pump drawn at same scale as Fig. 4L; H. Spermatheca drawn in Canada balsam; I. Spermathecae and genital fork drawn in phenol; J. Female wing; K. Male wing; L. Male terminalia; M. Female cibarium; same scale as Fig. 4N; N. Female cibarium. All figures after Young et al. (1983), except Fig. 4M, from California specimens. Scale in mm.

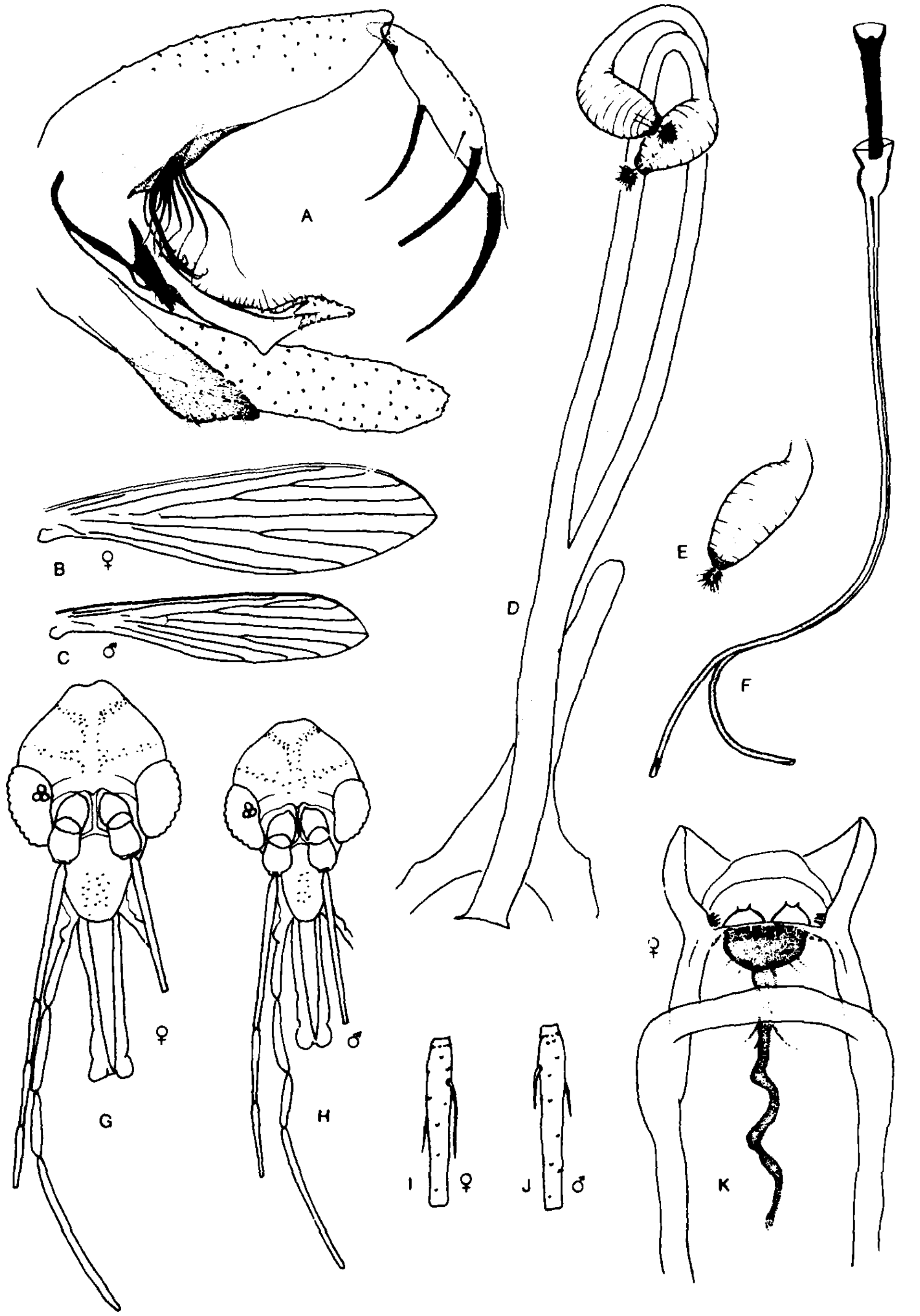

Fig. 5. *Lutzomyia aquilonia* (Fairchild and Harwood). A. Male terminalia; B. Female wing; C. Male wing; D. Spermathecae and genital fork; E. Spermatheca of allotype; F. Male aedeagal filaments, ejaculatory apodeme and sperm pump; G. Female head; H. Male head; I. Female flagellomere II; J. Male flagellomere II; K. Female cibarium. Drawn from Washington State specimens at same scale as comparable structures in Fig. 4.

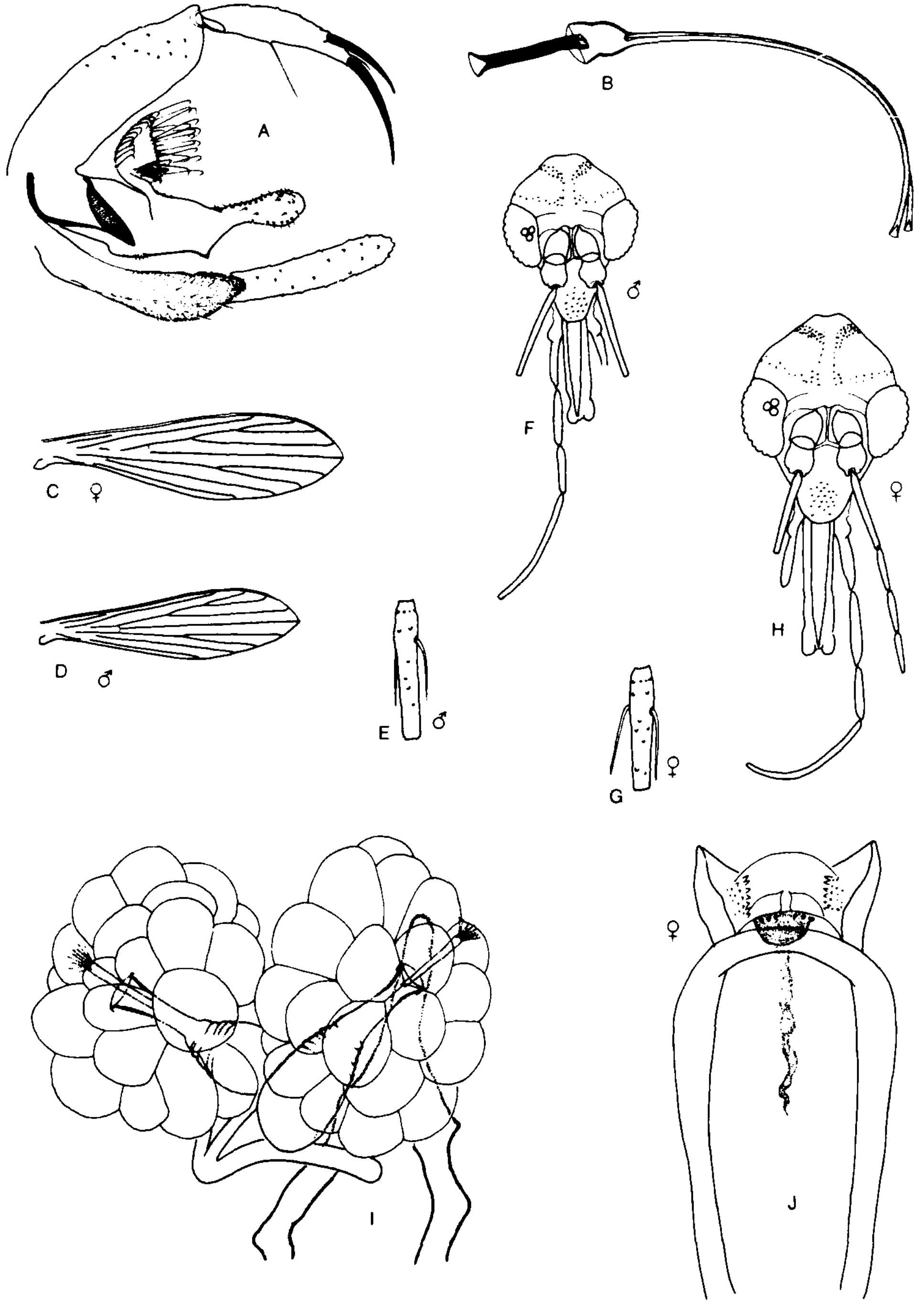

Fig. 6. *Lutzomyia anthophora* (Addis). A. Male terminalia; B. Male aedeagal filaments, ejaculatory apodeme and sperm pump; C. Female wing; D. Male wing; E. Male flagellomere II; F. Male head; G. Female flagellomere II; H. Female head; I. Spermathecae showing bubble-like evaginations and genital fork; J. Female cibarium. Drawn from Texas specimens at same scale as comparable structures in Fig. 4.

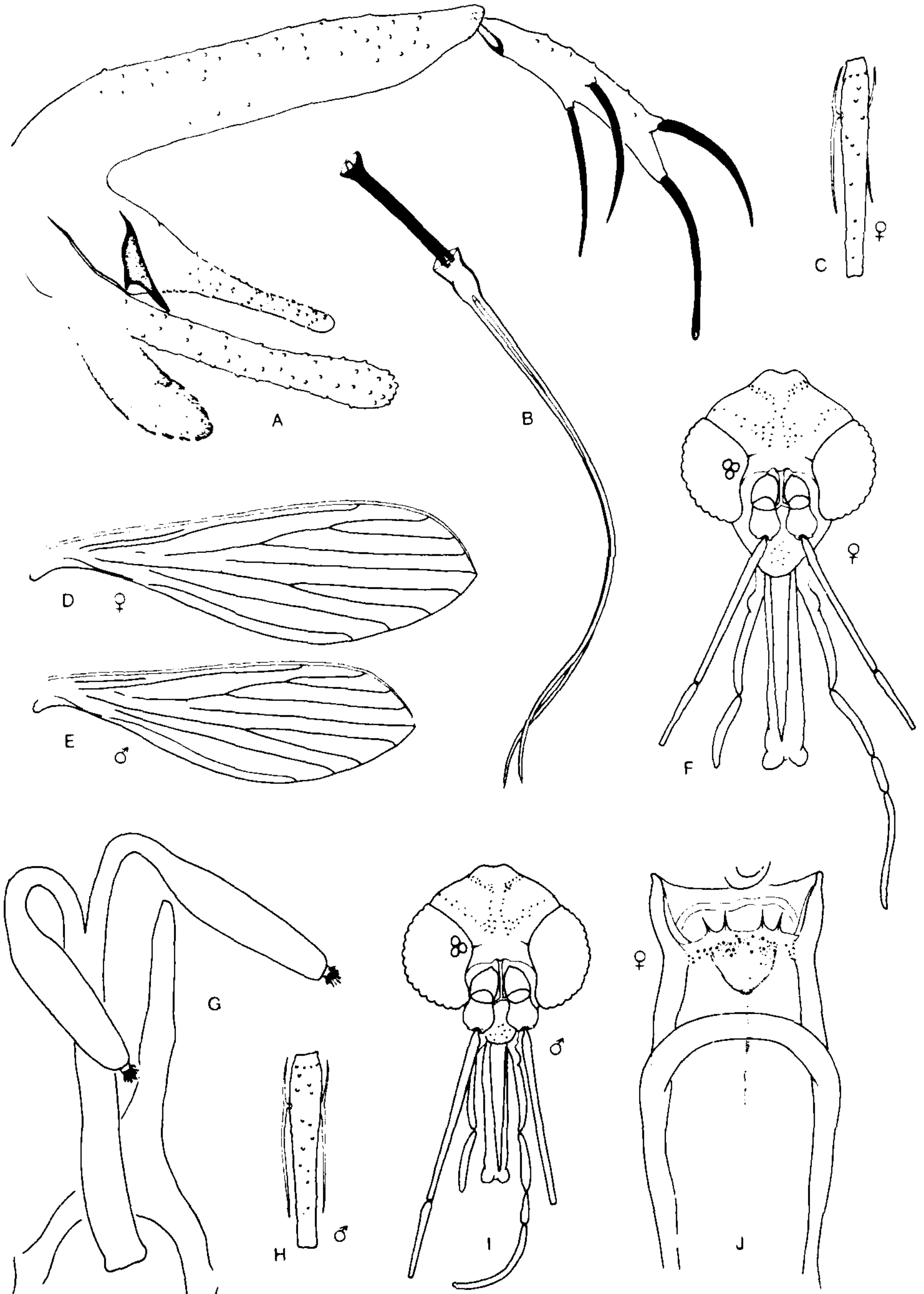

Fig. 7. *Lutzomyia shannoni* (Dyar). A. Male terminalia; B. Male aedeagal filaments, ejaculatory apodeme and sperm pump; C. Female flagellomere II; D. Female wing; E. Male wing; F. Female head; G. Spermathecae (thin outer envelope surrounding each spermatheca not shown) and genital fork; H. Male flagellomere II; I. Male head; J. Female cibarium. Drawn from Florida specimens at same scale as comparable structures in Fig. 4.

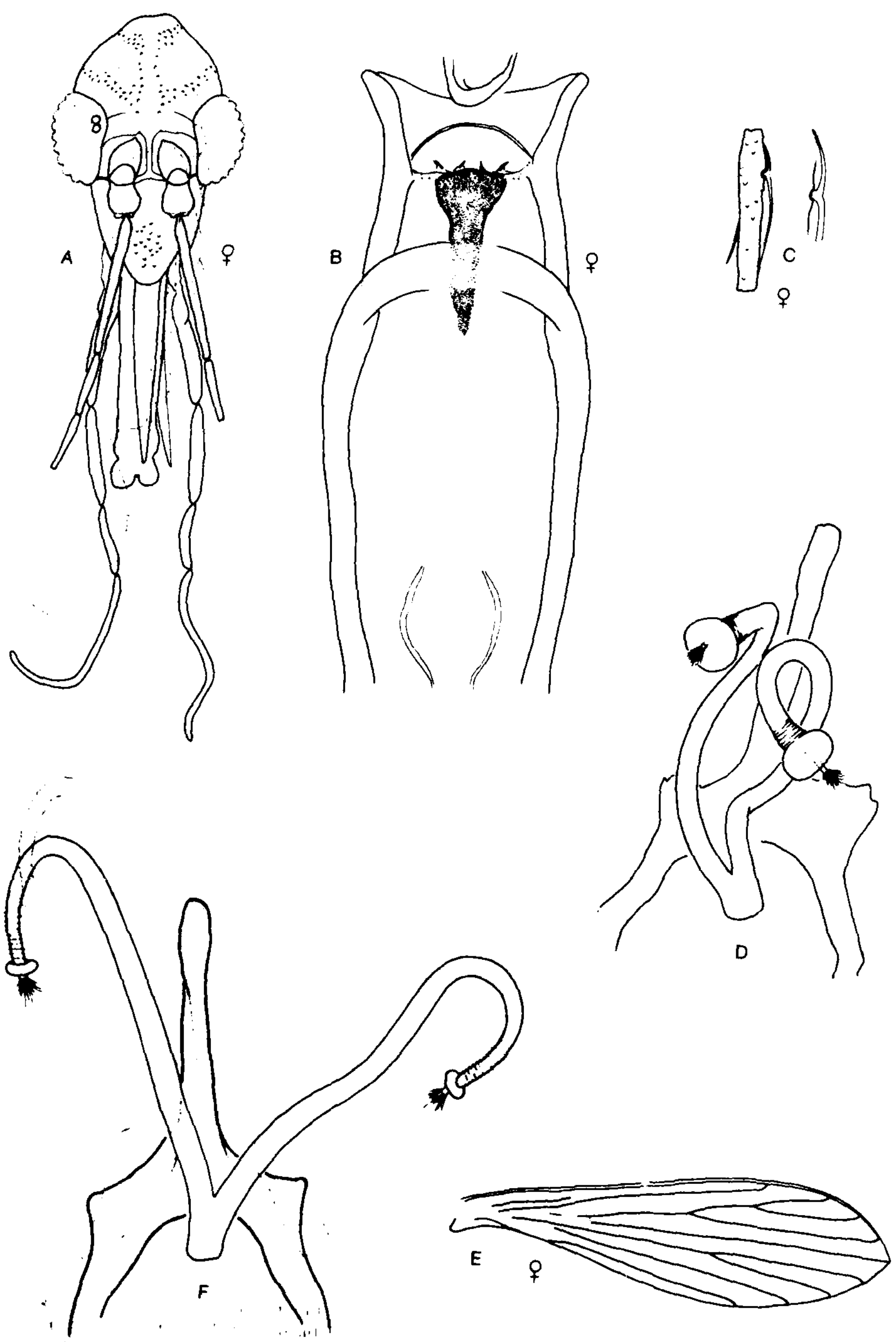

Fig. 8. *Lutzomyia tanyopsis* Young and Perkins n.sp. (8A-E holotype) and *Lutzomyia vexator* (Coquillett) from Ft. Ord, California (8F). A. Female head; B. Female cibarium; C. Female flagellomere II; D. Spermathecae and genital fork; E. Female wing; F. Spermathecae and genital fork. All structures drawn at same scale as those in Fig. 4.

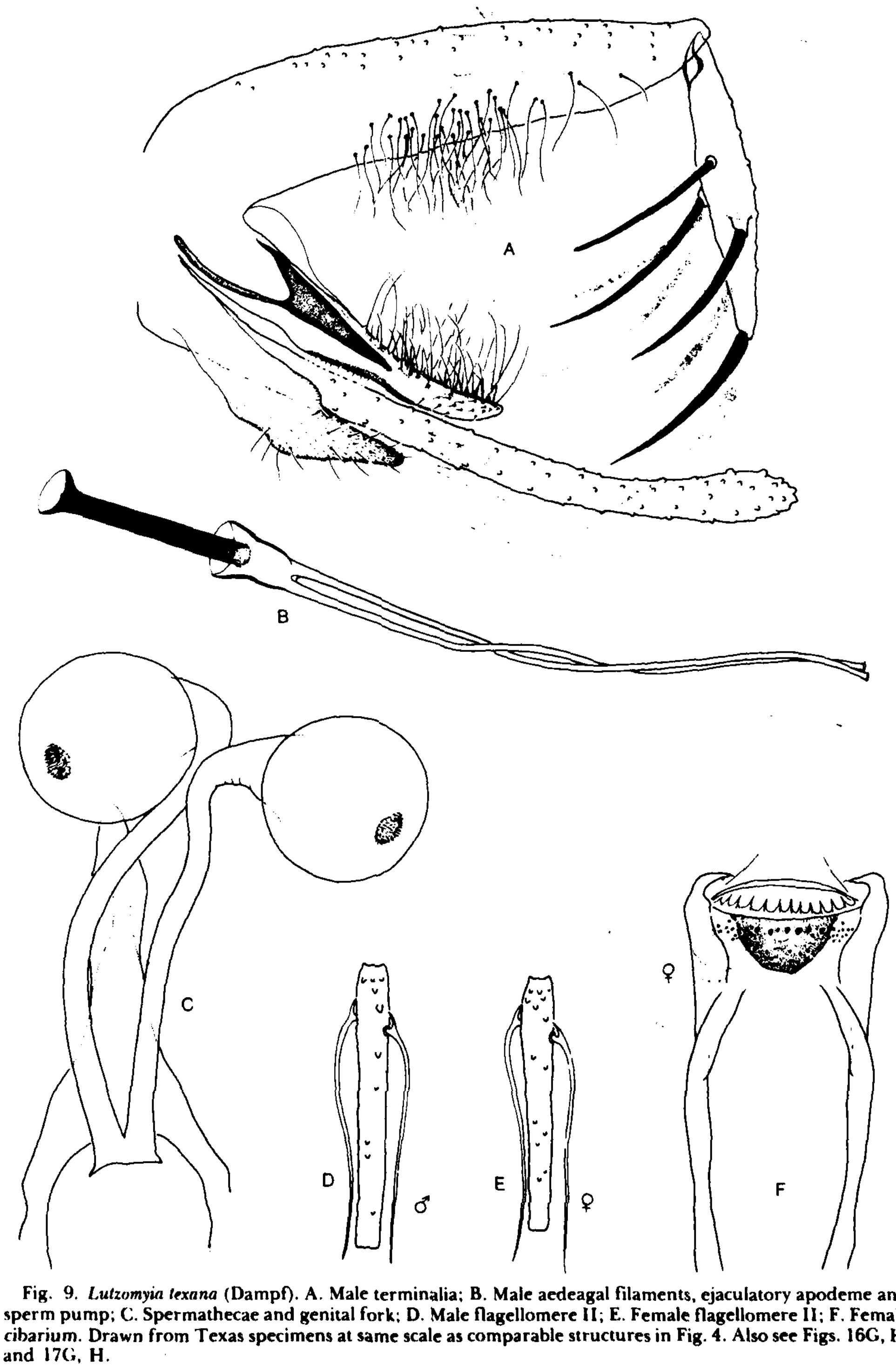

Fig. 9. *Lutzomyia texana* (Dampf). A. Male terminalia; B. Male aedeagal filaments, ejaculatory apodeme and sperm pump; C. Spermathecae and genital fork; D. Male flagellomere II; E. Female flagellomere II; F. Female cibarium. Drawn from Texas specimens at same scale as comparable structures in Fig. 4. Also see Figs. 16G, H, and 17G, H.

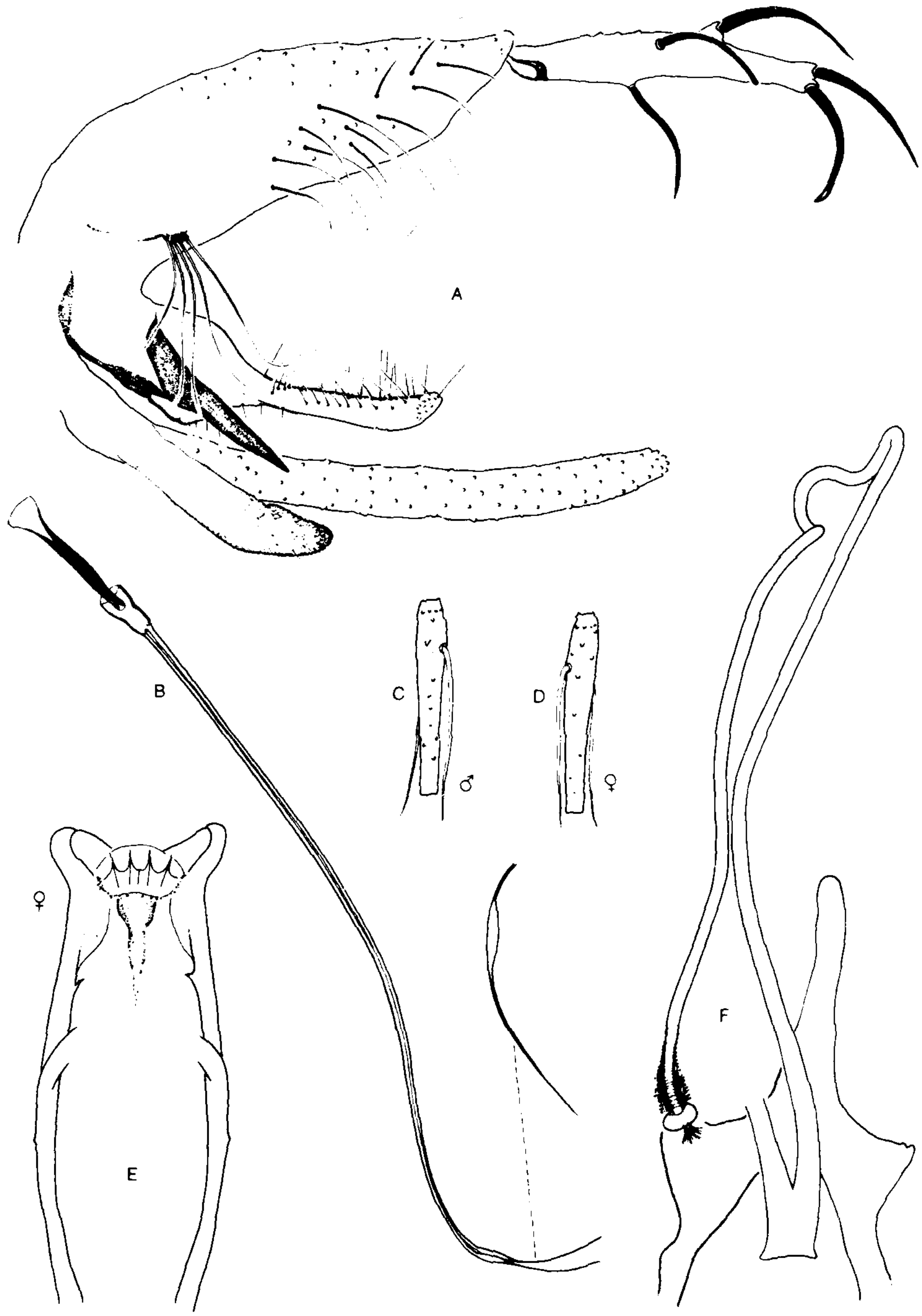

Fig. 10. *Lutzomyia apache* Young and Perkins n.sp. A. Male terminalia; B. Male aedeagal filaments, ejaculatory apodeme and sperm pump; C. Male flagellomere II; D. Female flagellomere II; E. Female cibarium; F. Spermatheca (only one is shown) and genital fork. Drawn from Springerville, Arizona, specimens at same scale as comparable structures in Fig. 4. Also see Figs. 16E, F and 17E, F.

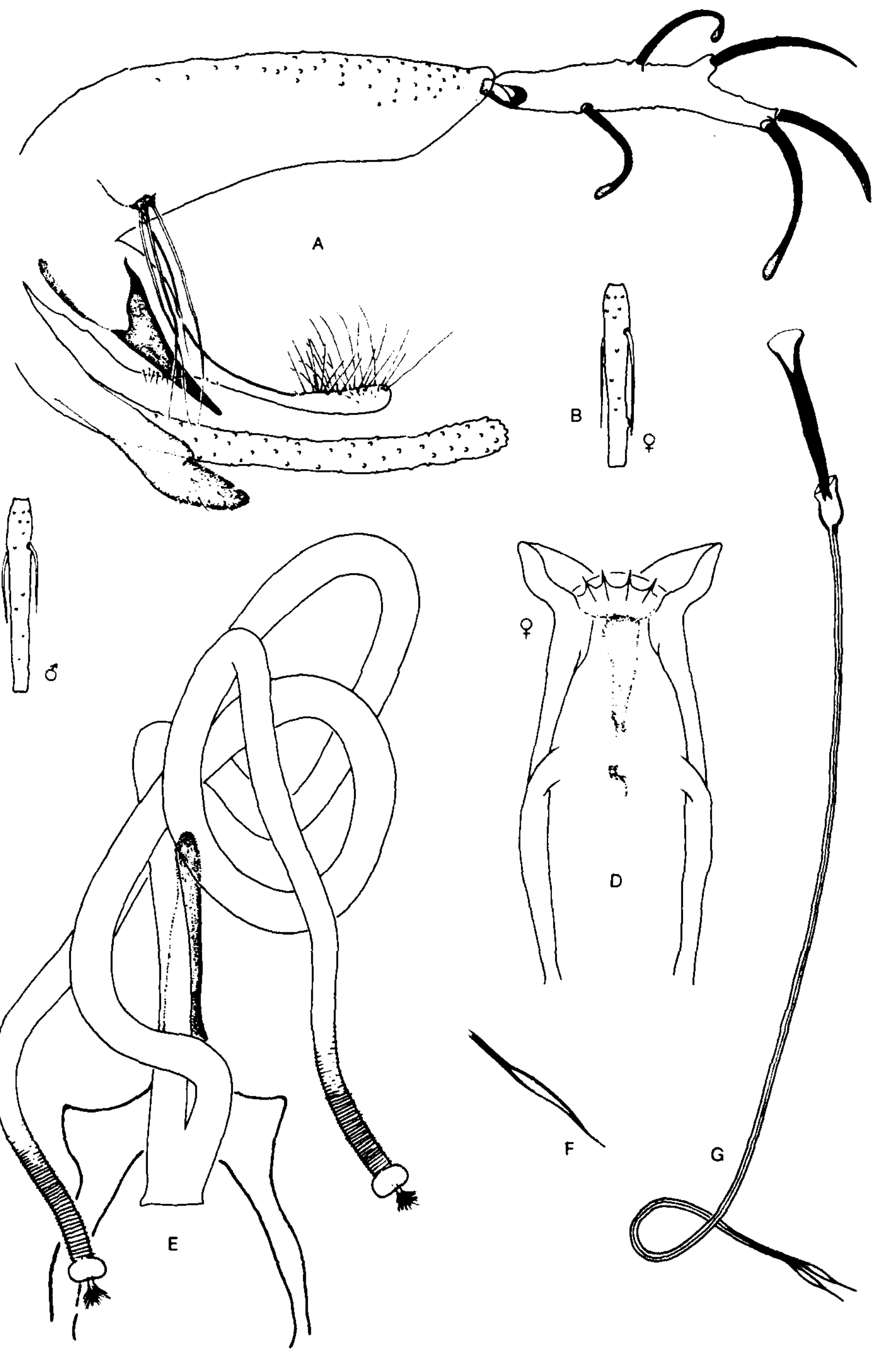

Fig. 11. *Lutzomyia oppidana* (Dampf). A. Male terminalia; B. Male flagellomere II; C. Female flagellomere II; D. Female cibarium; E. Spermathecae and genital fork; F. Tip of aedeagal filament; G. Aedeagal filaments, ejaculatory apodeme and sperm pump. Drawn from Mexico specimens at same scale as comparable structures in Fig. 4. Also see Figs. 16A, B and 17A, B.

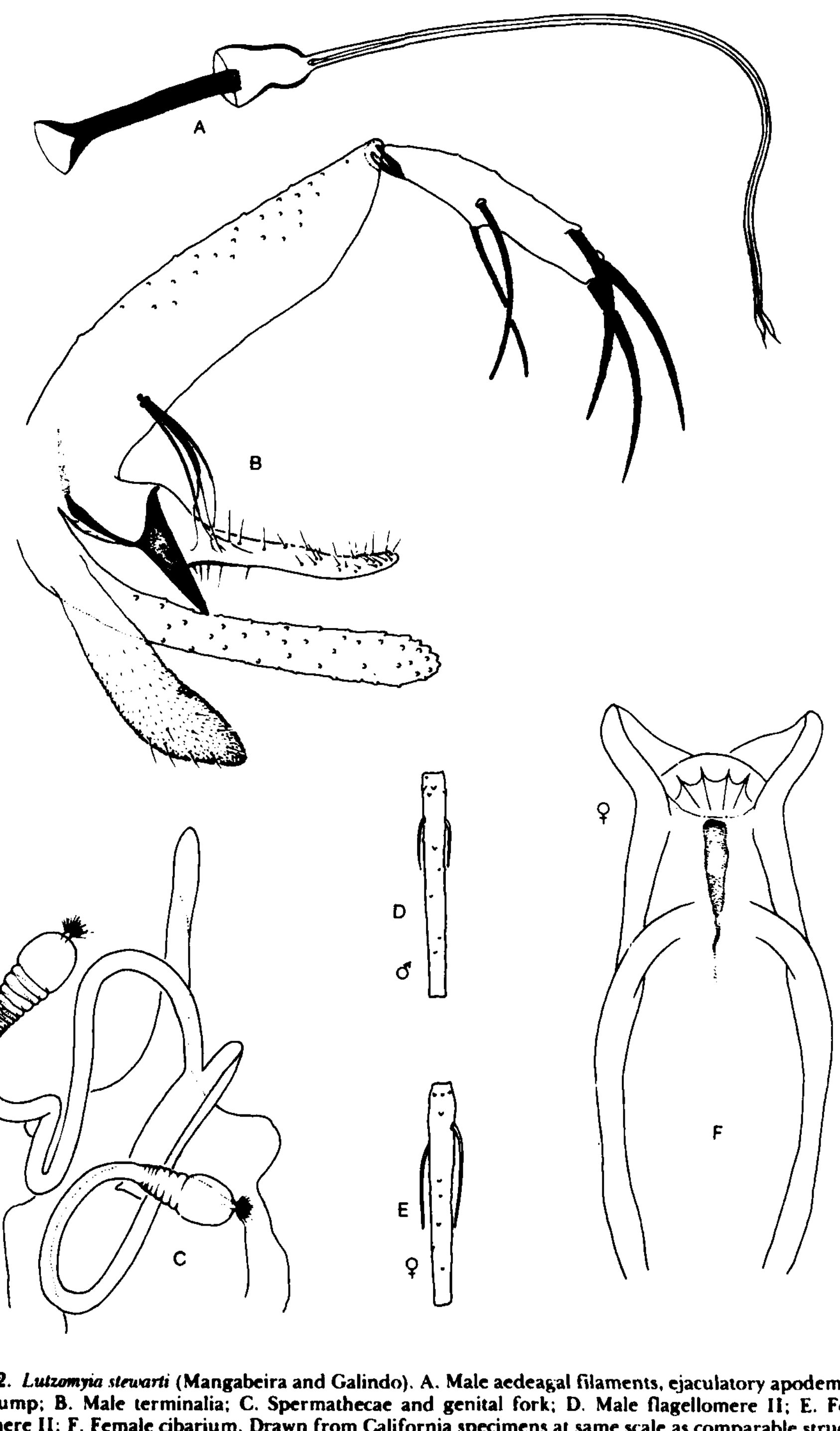

Fig. 12. *Lutzomyia stewarti* (Mangabeira and Galindo). A. Male aedeagal filaments, ejaculatory apodeme and sperm pump; B. Male terminalia; C. Spermathecae and genital fork; D. Male flagellomere II; E. Female flagellomere II; F. Female cibarium. Drawn from California specimens at same scale as comparable structures in Fig. 4. Also see Figs. 16C, D and 17C, D.

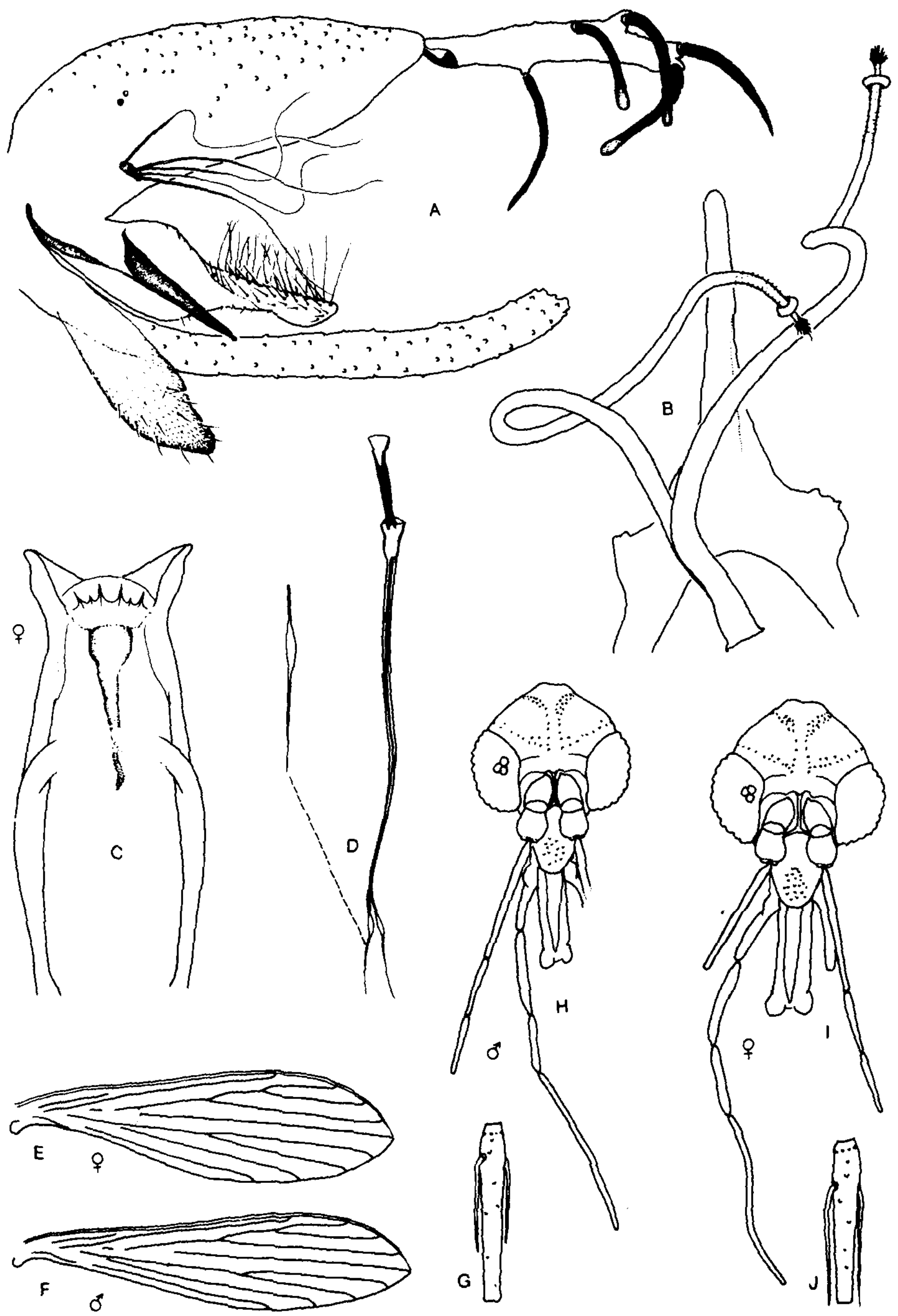

Fig. 13. *Lutzomyia vexator* (Coquillett). A. Male terminalia; B. Spermathecae and genital fork; C. Female cibarium; D. Male aedeagal filaments, ejaculatory apodeme and sperm pump with one filament tip enlarged; E. Female wing; F. Male wing; G. Male flagellomere II; H. Male head; I. Male flagellomere II; J. Female flagellomere II. Drawn from Florida specimens at same scale as comparable structures in Fig. 4. Also see Fig. 8F.

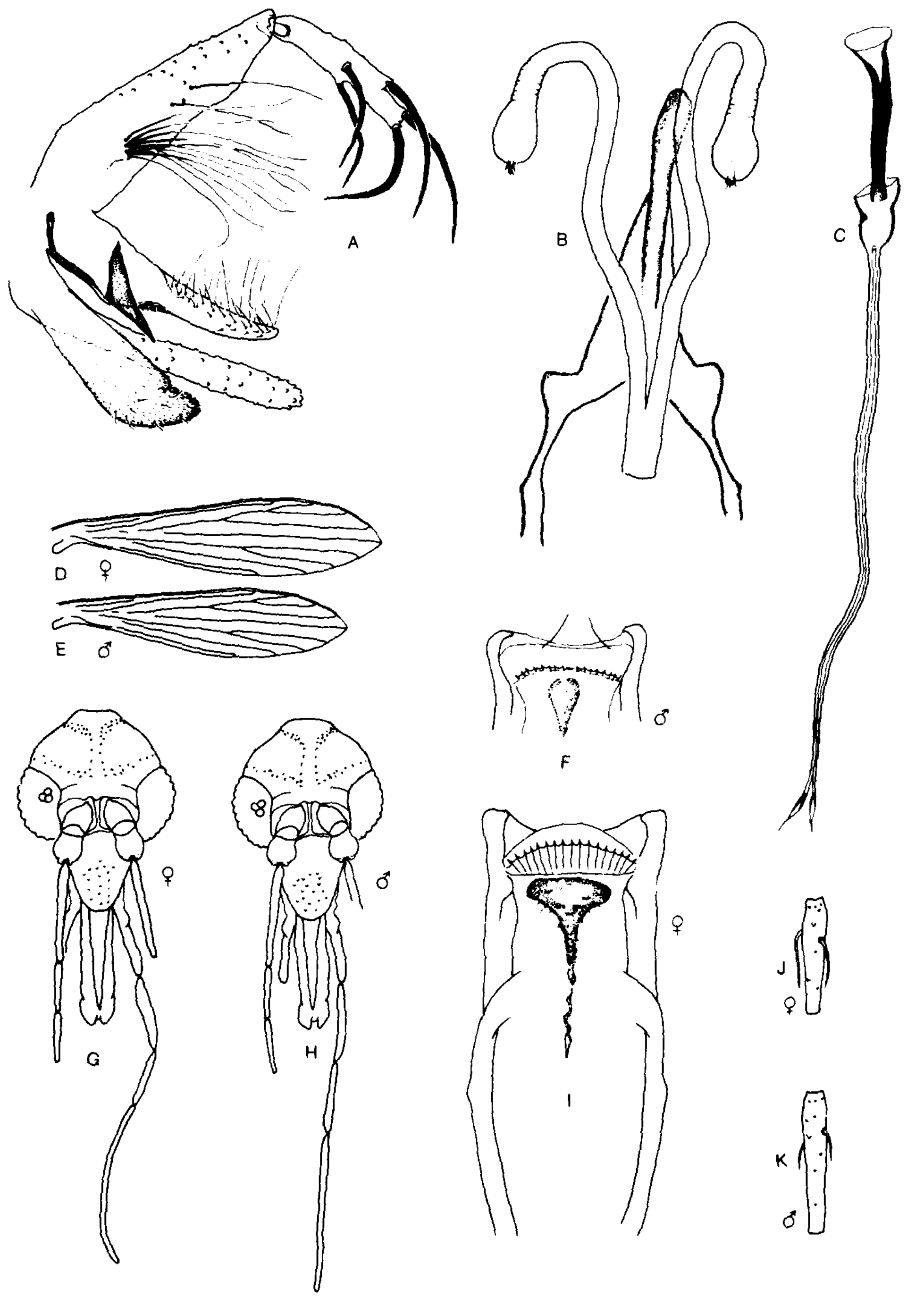

Fig. 14. *Lutzomyia californica* (Fairchild and Hertig). A. Male terminalia; B. Spermathecae and genital fork; C. Male aedeagal filaments, ejaculatory apodeme and sperm pump; D. Female wing; E. Male wing; F. Male cibarium; G. Female head; H. Male head; I. Female cibarium; J. Female flagellomere II; K. Male flagellomere II. Drawn from California specimens at same scale as comparable structures in Fig. 4.

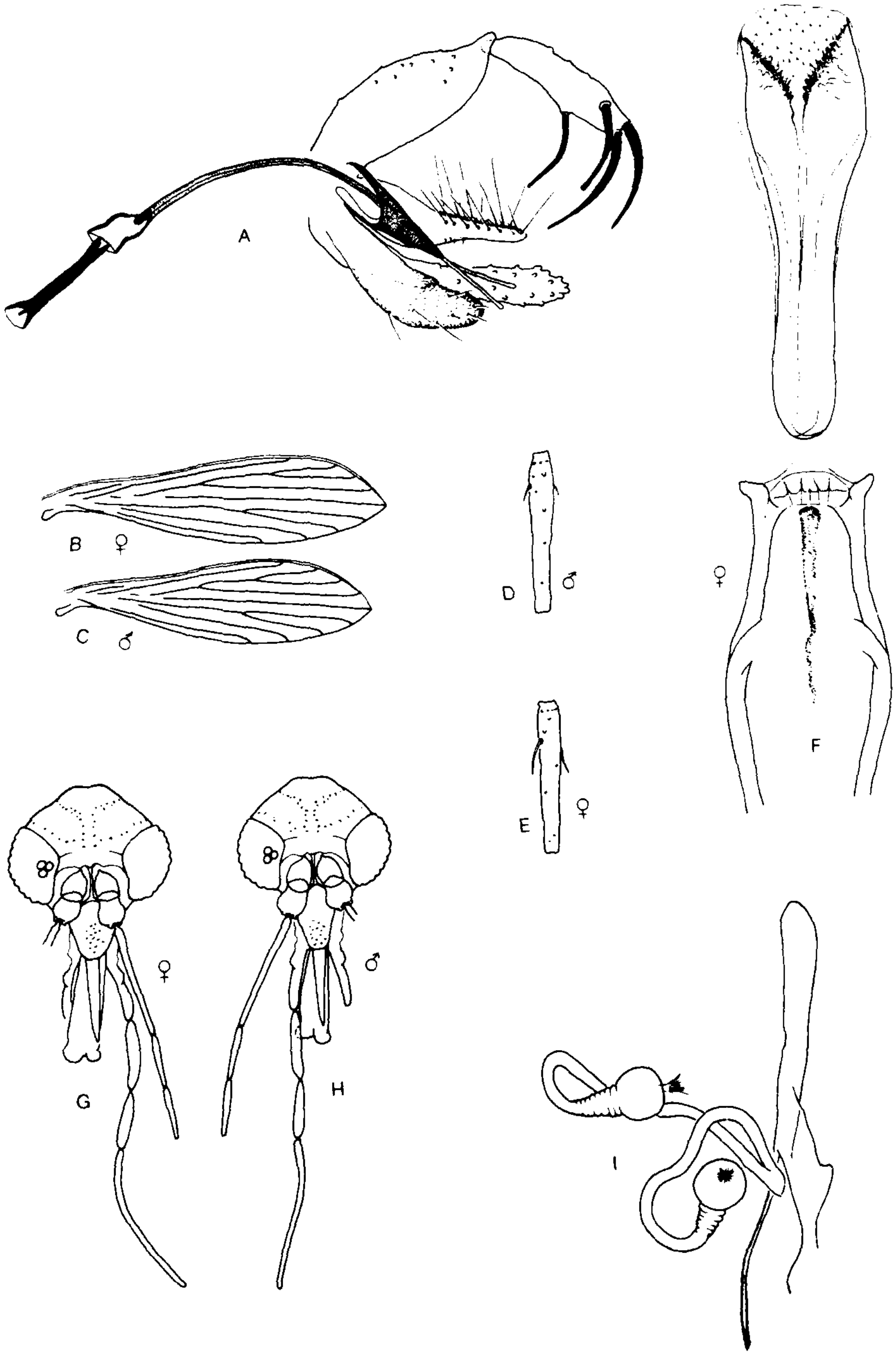

Fig. 15. *Lutzomyia cubensis* (Fairchild and Trapido). A. Male terminalia; B. Female wing; C. Male wing; D. Male flagellomere II; E. Female flagellomere II; F. Female cibarium and pharynx; G. Female head; H. Male head; I. Spermathecae and genital fork. Drawn from Florida specimens at same scale as comparable structures in Fig. 4.

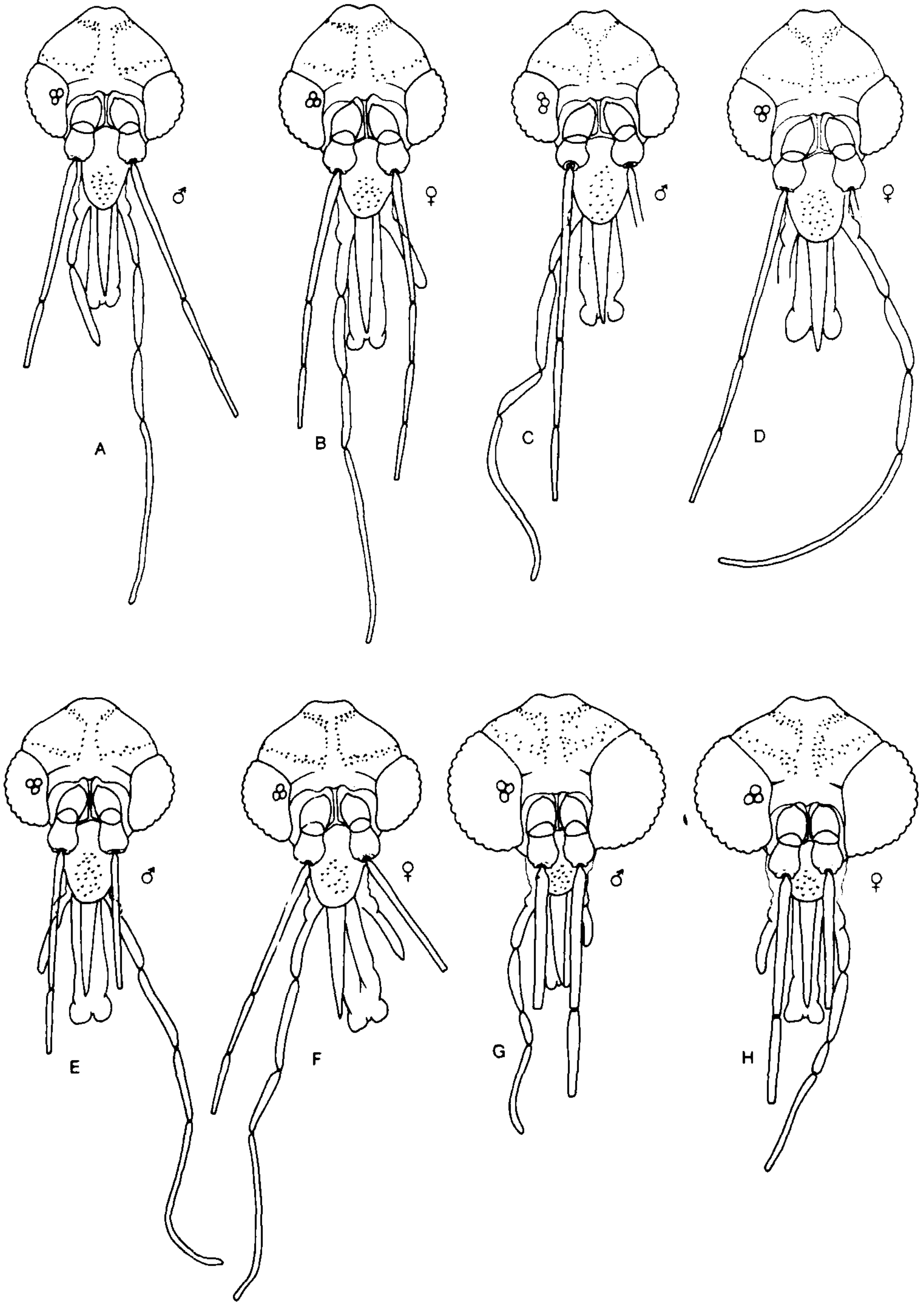

Fig. 16. Male and female heads of *Lutzomyia oppidana* (A and B); *L. stewarti* (C and D); *L. apache* (E and F), and *L. texana* (G and H). All drawn at same scale as heads in Fig. 4.

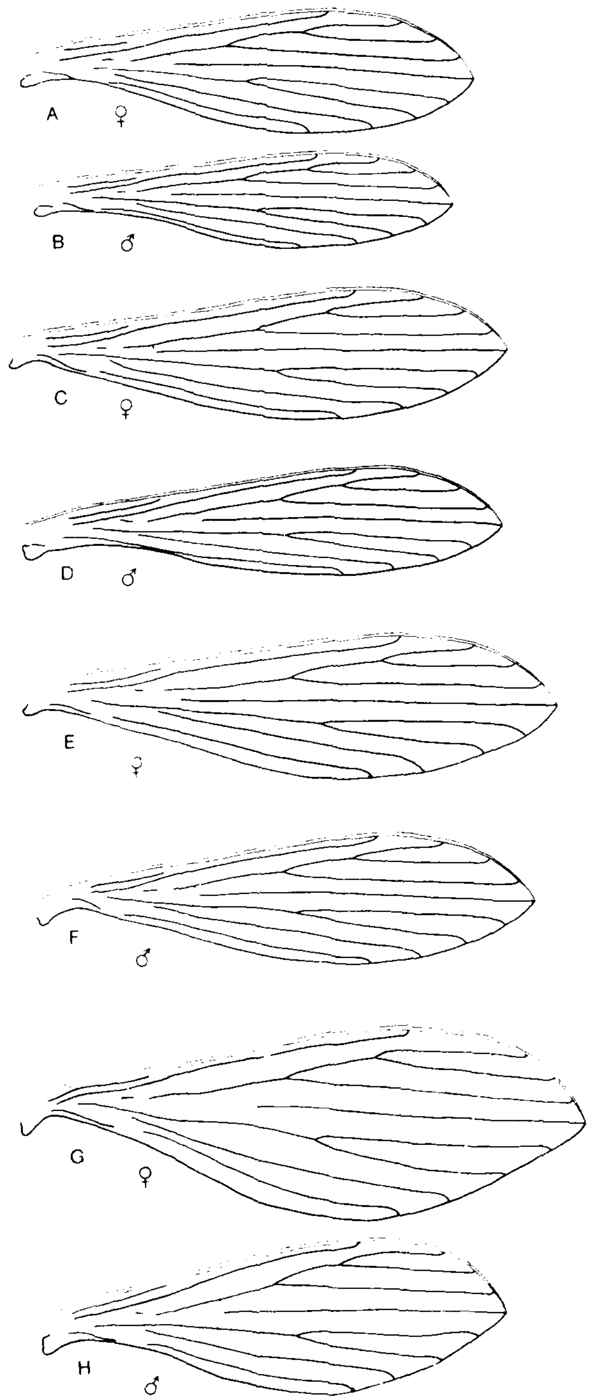

Fig. 17. Male and female wings of *Lutzomyia oppidana* (A and B); *L. stewarti* (C and D); *L. apache* (E and F), and *L. texana* (G and H). All drawn at same scale as wings in Fig. 4.

Fig. 18. Distribution of *Lutzomyia cruciata* and *L. diabolica* in North America.

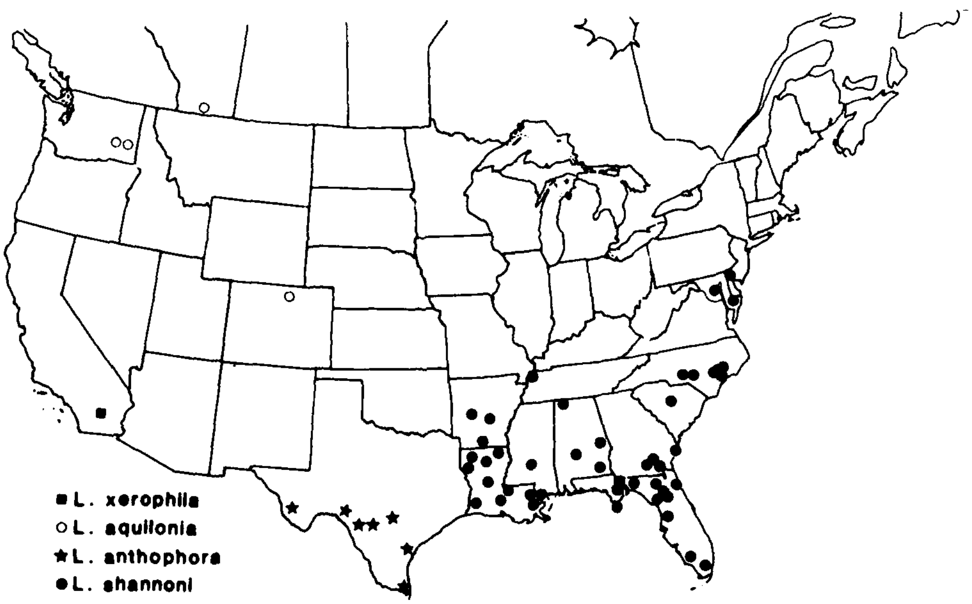

Fig. 19. Distribution of *Lutzomyia xerophila*, *L. aquilonia*, *L. anthophora* and *L. shannoni* in North America.

Fig. 20. Distribution of *Lutzomyia tanyopsis*, *L. texana* and *L. apache* in North America.

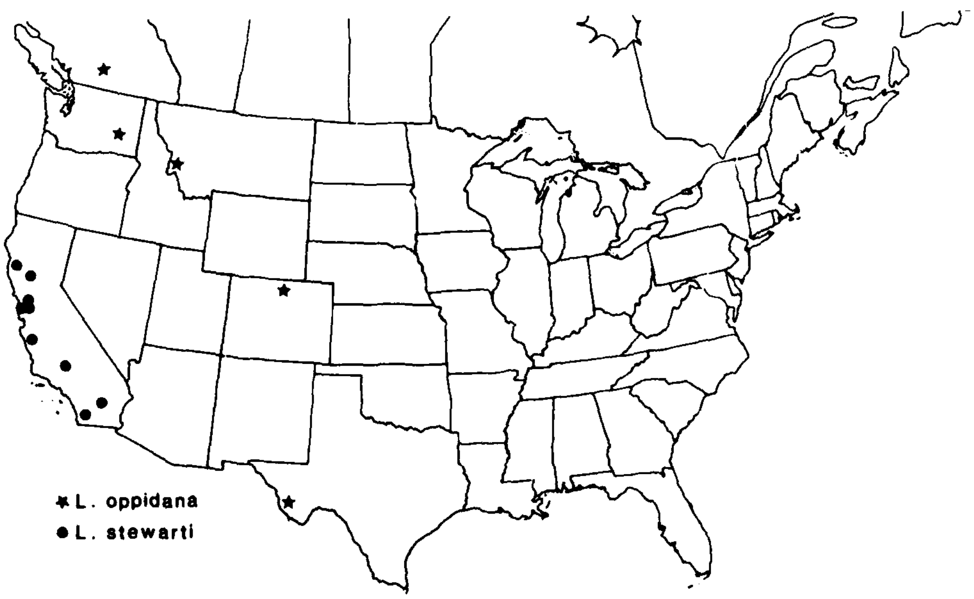

Fig. 21. Distribution of *Lutzomyia oppidana* and *L. stewarti* in North America.

Fig. 22. Distribution of *Lutzomyia vexator* in North America.

Fig. 23. Distribution of *Lutzomyia californica* and *L. cubensis* in North America.

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